Infection Prevention

Guidelines for Healthcare Facilities with Limited Resources

Linda Tietjen
Débora Bossemeyer
Noel McIntosh

JHPIEGO
An Affiliate of Johns Hopkins University
WORKING TO IMPROVE THE HEALTH OF WOMEN AND FAMILIES THROUGHOUT THE WORLD

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United States Agency for International Development

The authors have made every effort to check the accuracy of all information, the dosages of any drugs, and instructions for use of any devices or equipment. Because the science of infection prevention is rapidly advancing and the knowledge base continues to expand, readers are advised to check current product information provided by the manufacturer of:

- each drug to verify the recommended dose, method of administration and precautions for use; and
- each device, instrument or piece of equipment to verify recommendations for use and/or operating instructions.

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The **Training in Reproductive Health (TRH) Project** works globally to establish integrated (pre- and inservice) education and training systems to improve the performance of reproductive health professionals.

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This manual is a reference guide to infection prevention thinking and practices in hospitals and healthcare facilities where resources are limited, and in some countries (e.g., Nepal, Nigeria, Mozambique, Rwanda and Somalia) actually decreasing. It is based in large part on the experience gained during the last 11 years since publication of the first manual, *Infection Prevention for Family Planning Service Programs*[1]. It reflects what we (the authors) have learned from countless healthcare workers throughout the world who abstracted, translated, taught and used the simple, practical procedures and practices contained in that manual.

During the past decade, the success of the first manual as an international infection prevention reference for use in outpatient settings, such as family planning programs, has been amply documented. The challenge in writing this new manual has been to keep the content as simple and practical as possible while at the same time incorporating essential information on a much larger scale—infection prevention guidelines for hospitals providing general medical, surgical and obstetric services. Therefore, to make it as useful as possible, we sought input from a wide range of health professionals and international organizations, and we are deeply indebted to them for their interest, support and contributions. Specifically, the authors and JHPIEGO wish to thank:

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    - Pat Lynch has worked in the field of infection control since 1968. She was the first president of the US Association of Professionals in Infection Control and Epidemiology (APIC) in 1972, and is one of the original developers of the isolation system known as Body Substance Isolation that was incorporated into the Centers for Disease Control and Prevention's revised Guideline for Isolation Precautions in Hospitals in 1996. She also has consulted with practitioners in numerous hospitals with limited resources in the US and developing countries.

[1] Tietjen L, W Cronin and N McIntosh. 1992. *Infection Prevention for Family Planning Service Programs: A Problem-Solving Reference Manual.* Essential Medical Information Systems, Inc.: Durant, OK.

- Mark Davis is an experienced surgeon who now devotes much of his time to promoting safety in hospitals, the healthcare industry and manufacturers of medical and surgical safety products. His contribution to **Chapter 7** (**Safe Practices in the Operating Room**) and **Appendix D** (**Precautions for the Surgical Team**) was crucial and freely given. We also would like to acknowledge his consultation on JHPIEGO's new infection prevention video, *Safe Practices in the Operating Room*, which is an integral part of the learning materials used in training health workers.

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PREFACE

Since publication of the first manual 11 years ago, considerable progress has been made globally in understanding the basic principles of infection prevention as well as acceptance and use of evidence-based infection prevention practices. Key concepts and practices that are now more widely accepted in many countries include:

- Recognition of the dual role of infection prevention not only in reducing the risk of disease transmission to clients and patients but also protecting healthcare workers at all levels—from physicians and nurses to cleaning and housekeeping staff.
- Role of handwashing in preventing disease transmission and, where clean water is not readily available, the use of inexpensive, easy-to-make, alcohol-based antiseptic handrubs.
- Importance of first decontaminating all soiled instruments, needles and syringes and other items with dilute (0.5%) bleach solution if cleaning (washing and rinsing) is done by hand.
- Need for thorough cleaning of soiled instruments, gloves and other items if final processing, either by high-level disinfection (HLD) or sterilization, is to be effective.
- Use of HLD by boiling or steaming as cost-effective, readily available and acceptable alternative to sterilization (autoclaving or dry heat) for most surgical procedures.
- Multiple uses of dilute chlorine solutions made from inexpensive, commercial bleach (sodium hypochlorite) products for:
  - decontamination (0.5%) of soiled surgical instruments as well as cleaning large surfaces (examination tables),
  - HLD (0.1%) of surgical instruments and other items, and
  - preparation of safe drinking water (0.001%).
- Use of the "no touch" surgical technique when performing procedures, such as IUD insertion and removal or vacuum aspiration for incomplete abortion, thereby allowing examination gloves to be safely substituted for sterile surgical gloves, which are expensive and often difficult to obtain.

Because of the demand for infection prevention guidelines for use at district hospitals, not just ambulatory family planning services, this manual contains many new chapters and has been completely rewritten to take advantage of the wealth of new information and practical interventions. The content, however, is not all encompassing, nor is it encyclopedic. The intent is to provide the user a quick reference to what

the essentials are without having to consult other sources. In addition, the manual has been designed to provide the information and recommendations in a simple, easily understandable format so that users can find what they want, when they want it.

The infection prevention principles and scientific information, on which this manual is based, are universally applicable. In selecting the material, the emphasis has been on choosing those practices and procedures that are doable even in the poorest settings. Ones designed to minimize cost and the need for expensive technology or fragile equipment while at the same time assuring a high degree of safety. As such, this manual is not intended to be a major resource for infection prevention programs in affluent settings. In fact, some of the practices recommended may be at odds with established norms; for example, the need for decontamination as the first step in processing soiled instruments and other items in order to make them safer for cleaning staff to handle; or an even larger issue—the reuse of disposable (single use) items.

Because of the severe cost constraints faced by hospital managers in the poorest countries, the manual is geared to prevention, especially preventing postoperative obstetrical and general surgical infections, as well as those resulting from the use of invasive medical devices. Infection surveillance and control, both important elements of infection control programs, are only briefly touched on because sound surveillance systems are lacking in most countries and resources to treat hospital-acquired (nosocomial) infections or antibiotic-resistant infections, even when identified, rarely are available.[1]

HOW TO USE THE MANUAL

A key purpose of the manual is to enable hospital administrators, clinic managers and healthcare professionals working in limited resource settings to develop their own uniform infection prevention policies and service delivery guidelines. It is recognized, however, that the strategies, priorities and proven methods of infection risk reduction described in this manual will need to be adapted to reflect the existing conditions in each country. Only through this process can much needed changes be implemented and patient care in hospitals and clinics improved.

Content and Organization

The material in this manual is divided into four parts. In the first part, **FUNDAMENTALS OF INFECTION PREVENTION,** basic principles and the recommended practices of modern infection prevention programs

[1] In 1997 a second manual dealing with the special infection prevention needs of developing countries became available. In addition to providing new insights on improving infection prevention, it also contains much needed practical guidance on the role of infection surveillance and control efforts when resources are limited. Moreover, it provides a broader framework, one that includes the control and treatment of antibiotic-resistant nosocomial infections. Readers are encouraged to consult this manual for additional information on these topics. (Lynch P et al. 1997. *Infection Prevention with Limited Resources*: *A Handbook for Infection Committees.* ETNA Communications: Chicago.)

are described. The emphasis is on providing the scientific data supporting their use and appropriateness in situations where resources and manpower are limited. Also, several new chapters have been added based on the new Standard Precautions, which must be used when caring for **all** clients and patients attending healthcare facilities. Introduced in 1996 by CDC, Standard Precautions are the first level of the revised isolation guidelines that replace the older Universal Precautions and Body Substance Isolation Precautions. Moreover, because the most serious and frequent site for accidental injuries and exposure to bloodborne pathogens is the operating room, a separate chapter and appendix detailing safety practices, tips on how to design safer operations and a full set of safety checklists for making the operating room safer have been added.

The second part, **PROCESSING INSTRUMENTS, GLOVES AND OTHER ITEMS**, has been expanded in order to incorporate additional, but essential information, needed by healthcare staff working in district hospitals. When combined with data from several new or updated appendices, which contain more detailed supplemental "how to" information, health workers now have the information they need to solve many of the instrument and equipment problems and reprocessing issues not previously addressed.

IMPLEMENTING INFECTION PREVENTION IN HEALTHCARE FACILITIES, the third part, focuses on coordinating and managing the special infection prevention needs and services at district-level hospitals, where the volume and type of health services offered are greater than in the ambulatory setting. In hospitals, housekeeping services and traffic flow systems and activity patterns are more diverse and complex as well. Moreover, the risk of exposure to bloodborne pathogens and other life-threatening infections is not confined just to operating and recovery rooms and patient care areas. Staff working in routine chemistry, clinical pathology and bacteriology laboratories as well as those providing blood bank and transfusions services need to be aware of the risks and how to prevent accidental injuries and exposures. Therefore, guidelines and recommended preventive practices for these staff have been included. Finally, in dealing with the overall management of infection prevention programs, the role of the infection prevention committee or working group is critical for handling routine problems, developing workable guidelines and protocols, actively supporting their use and modeling the appropriate preventive behaviors. Representatives from all parts of the healthcare facility who are interested in making the workplace safer should be encouraged to serve this vitally important function.

The final part, **NOSOCOMIAL INFECTIONS** is all new. The magnitude of the HIV/AIDS crisis globally, coupled with re-emergence of tuberculosis, especially multidrug resistant strains, has changed the way healthcare is provided. Hospitals now need practical, symptom-based isolation guidelines to prevent patients and health workers at all levels

from being inadvertently exposed to these serious infectious diseases as well as others transmitted by the airborne, droplet and contact routes. Therefore, specific information and detailed guidance is provided on how to implement and use the second level of the CDC isolation guidelines, Transmission Precautions for Hospitalized Patients. Also included is practical guidance designed to help prevent the most common and serious nosocomial infections in hospitalized patients—urinary tract infections, diarrhea and pneumonia—as well as infections following surgery, maternal and newborn infections and those associated with the use of an ever-increasing number of intravascular devices. Because safely managing food and water in hospitals is important in preventing the spread of infections, these topics are also covered. Finally, because outbreaks of serious infections do occur, guidelines are included for how to investigate them as well as how to monitor infection prevention program activities most cost-effectively.

Using the Manual

It is anticipated this manual will serve as an international reference guide for use in limited resource settings. Moreover, we hope that health educators and trainers, public health and medical officials, and hospital managers as well as lay groups will find the information, practices and processes relevant and easy to use in adapting or developing their own infection prevention policies, guidelines, norms, education and training materials and healthcare monitoring tools. The content also may be used in different ways including:

- as a text for preservice education, group-based training or on-the-job learning programs; or
- as content for developing teaching, job or behavior change aids.

For each of these uses, the content may be produced and distributed in a variety of formats (paper-based, CD-ROM or via Internet). Finally, to facilitate the manual's adaptation and use, each chapter has a set of learning objectives, is fully referenced and is page numbered by chapter. Thus, each chapter can be reprinted as a stand-alone document for use as a handout when giving presentations.

ONE

INTRODUCTION TO INFECTION PREVENTION

KEY CONCEPTS you will learn in this chapter include:

- What the basic principles of infection prevention are
- What conditions allow infections to be transmitted to others
- How to stop the spread of infectious diseases
- What the role of the CDC isolation guidelines is in preventing nosocomial infections

BACKGROUND

People receiving health and medical care, whether in a hospital or clinic, are at risk of becoming infected unless precautions are taken to prevent infection. Nosocomial (hospital-acquired) infections are a significant problem throughout the world and are increasing (Alvarado 2000). For example, nosocomial infection rates range from as low as 1% in a few countries in Europe and the Americas to more then 40% in parts of Asia, Latin America and sub-Saharan Africa (Lynch et al 1997).

Most of these infections can be prevented with readily available, relatively inexpensive strategies by:

- adhering to recommended infection prevention practices, especially hand hygiene and wearing gloves;
- paying attention to well-established processes for decontamination and cleaning of soiled instruments and other items, followed by either sterilization or high-level disinfection; and
- improving safety in operating rooms and other high-risk areas where the most serious and frequent injuries and exposures to infectious agents occur.

How Risky is Working in a Hospital or Health Clinic

Healthcare workers, including support staff (e.g., housekeeping and maintenance and laboratory personnel), who work in these settings also are at risk of exposure to serious, potentially life-threatening infections. For example, in the US, more than 800,000 needlestick injuries occur each year despite continuing education and vigorous efforts aimed at preventing such accidents (Rogers 1997), including:

- reducing unnecessary and unsafe injections,
- training all staff to immediately dispose of needles and syringes in sharps containers without recapping—attempting to recap them accounts for one third of all needlesticks (Jagger et al 1988),[1]
- placing disposable sharps containers within arm's reach if possible, and
- increasing use of needleless injection systems and shielded syringes.

In many developing countries, however, the risk of needlestick injuries and accidental exposure to blood or body fluids is even higher (Phipps et al 2002). Moreover, because introduction of needleless injection systems is not feasible in countries with limited resources, it is important that healthcare staff **know** and **use** recommended infection prevention practices to minimize their risk of accidental exposure or injury (Tietjen 1997).

Purpose of This Chapter

The **purpose** of this chapter is to assist healthcare workers and hospital and clinic supervisors, managers and administrators understand the basic principles of infection prevention and recommended processes and practices. Also presented is an overview of the Centers for Disease Control and Prevention (CDC) isolation precaution guidelines for hospitals (Garner and HICPAC 1996). These guidelines replace both Universal Precautions and Body Substance Isolation Precautions and provide the framework on which **Part 1. Fundamentals of Infection Prevention** and **Part 2. Processing Instruments, Gloves and Other Items** are based.

DEFINITIONS

The terms **asepsis (aseptic technique)**, **antisepsis**, **decontamination**, **cleaning**, **high-level disinfection** and **sterilization** often are confusing. For the purposes of these guidelines, the following definitions will be used:

- **Asepsis** and **aseptic technique**. Combination of efforts made to prevent entry of microorganisms into any area of the body where they are likely to cause infection. The goal of asepsis is to **reduce to a safe level**, or **eliminate**, the number of microorganisms on both animate (living) surfaces (skin and mucous membranes) and inanimate objects (surgical instruments and other items).
- **Antisepsis**. Process of reducing the number of microorganisms on skin, mucous membranes or other body tissue by applying an antimicrobial (antiseptic) agent.
- **Decontamination**. Process that makes inanimate objects **safer** to be handled by staff **before** cleaning (i.e., inactivates HBV, HCV and HIV and reduces, but does not eliminate, the number of other contaminating microorganisms).

[1] If recapping must be done, health workers should be trained in the one-hand technique (see **Chapter 7**).

Ideally, soiled surgical instruments, gloves and other items should always be handled by staff wearing gloves or using forceps. Because this is not always possible, it is safer first to soak these soiled items for 10 minutes in 0.5% chlorine solution, especially if they will be cleaned by hand (Nyström 1981). Metal objects should then be rinsed to prevent corrosion before cleaning (Lynch et al 1997). Other objects that should be decontaminated, by wiping with the 0.5% chlorine solution, include large surfaces (e.g., pelvic examination or operating tables) and equipment that come in contact with patients' blood or body fluids, secretions or excretions (except sweat).

- **Cleaning**. Process that physically removes all visible dust, soil, blood or other body fluids from inanimate objects as well as removing sufficient numbers of microorganisms to reduce risks for those who touch the skin or handle the object. (It consists of thoroughly washing with soap or detergent and water, rinsing with clean water and drying.[2])
- **High-level disinfection (HLD)**. The process that eliminates **all** microorganisms **except some** bacterial endospores from inanimate objects by boiling, steaming or the use of chemical disinfectants.
- **Sterilization**. Process that eliminates **all** microorganisms (bacteria, viruses, fungi and parasites) **including** bacterial endospores from inanimate objects by high-pressure steam (autoclave), dry heat (oven), chemical sterilants or radiation.

IMPORTANT CONCEPTS

Microorganisms are the causative agents of infection. They include bacteria, viruses, fungi and parasites. For infection prevention purposes, bacteria can be further divided into three categories: vegetative (e.g., staphylococcus), mycobacteria (e.g., tuberculosis) and endospores (e.g., tetanus). Of all the common infectious agents, endospores are difficult to kill due to their protective coating.[3]

Colonization means that pathogenic (illness or disease causing) organisms are present in a person (i.e., they can be detected by cultures or other tests) but are not causing symptoms or clinical findings (i.e., cellular changes or damage). **Infection** means that the colonizing organisms now are causing an illness or disease (cellular response) in the person. Coming in contact with and acquiring new organisms, while increasing the risk of infection, usually does not lead to infection because the body's natural defense mechanisms, including the immune system, are able to tolerate and/or destroy them. Thus, when organisms are transmitted from one person to another, colonization

[2] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

[3] Prions, which are protein-containing infectious agents present in brain, spinal column and eye tissue of patients with Creutzfeldt-Jakob disease, are even harder to kill (see **Chapter 11**).

rather than infection generally is the result. Colonized persons, however, can be a major source of transfer of pathogens to other persons (cross-contamination), especially if the organisms persist in the person (chronic carrier), such as with HBV, HCV and HIV.

Infection prevention largely depends on placing barriers between a **susceptible host** (person lacking effective natural or acquired protection) and the microorganisms. **Protective barriers** are physical, mechanical or chemical processes that help prevent the spread of infectious microorganisms from:

- person to person (patient, healthcare client or health worker); and/or
- equipment, instruments and environmental surfaces to people.

WHICH PROCESS TO USE

In 1968, Spaulding proposed three categories of potential infection risk to serve as the basis for selecting the prevention practice or process to use (e.g., sterilization of medical instruments, gloves and other items) when caring for patients. This classification has stood the test of time and still serves as a good basis for setting priorities for any infection prevention program. The Spaulding categories are summarized below:

- **Critical**. These items and practices affect normally sterile tissues or the blood system and represent the highest level of infection risk. Failure to provide management of sterile or, where appropriate, high-level disinfected items (e.g., surgical instruments and gloves), is most likely to result in infections that are the most serious.

- **Semicritical**. These items and practices are second in importance and affect mucous membranes and small areas of nonintact skin. Management needs are considerable and require knowledge and skills in:

  - handling many invasive devices (e.g., gastrointestinal endoscopes and vaginal specula),
  - performing decontamination, cleaning and high-level disinfection, and
  - gloving for personnel who touch mucous membranes and nonintact skin.

- **Noncritical**. Management of items and practices that involve intact skin and represent the lowest level of risk. Some (e.g., hand hygiene) are more important than others. Poor management of noncritical items, such as overuse of examination gloves, often consumes a major share of resources while providing only limited benefit.

Instrument Processing

Decontamination is the first step in processing soiled (contaminated) surgical instruments, gloves and other items, especially if they will be cleaned by hand (Nyström 1981). For example, briefly soaking contaminated items in 0.5% chlorine solution, or other locally available disinfectants, rapidly kills HBV[4] and HIV, thereby making the instruments and other items safer to be handled during cleaning (AORN 1990; DHMH 1990; Lynch et al 1997). Larger surfaces, such as examination and operating tables, laboratory bench tops and other equipment that may have come in contact with blood or other body fluids also should be decontaminated. Wiping with a suitable disinfectant (e.g., 0.5% chlorine solution or 1–2% phenol) is a practical, inexpensive way to decontaminate them.

After instruments and other items have been decontaminated, they need to be cleaned and finally either sterilized or high-level disinfected (Lynch 1997; Rutala 1993; Tietjen and McIntosh 1989). As outlined in **Table 1-1,** the process selected for final processing depends on whether the items will touch intact mucous membranes or broken skin or tissue beneath the skin that normally is sterile (Spaulding 1968).

Table 1-1. Final Processing for Surgical Instruments, Gloves and Other Items

TISSUE	FINAL PROCESSING	EXAMPLES
Intact mucous membranes or broken skin	**High-level disinfection** (HLD) destroys all microorganisms except some endospores.[a]	Uterine sounds, vaginal specula and plastic cannulae for suction curettage
Blood stream or tissue beneath the skin which normally is sterile	**Sterilization** destroys all microorganisms, including endospores.	Surgical instruments such as scalpels, trocars for insertion/removal of Norplant® implants and surgical gloves

[a] Bacterial endospores are forms of bacteria that are very difficult to kill because of their coating. Types of bacteria that make endospores include those causing tetanus (*Clostridium tetani*), gangrene (*Clostridium perfringens*) or anthrax (*Bacillus anthracis*).

Adapted from: Spaulding 1968.

When Is Sterilization Absolutely Essential? When Is HLD an Acceptable Alternative?

Most authorities recommend sterilization as the final step in processing instruments and other items used for surgical procedures. Some guidelines, however, are more flexible, and state that when sterilization equipment is **not** available, HLD can be used. In fact, the use of sterilization is not possible or practical in certain situations (Rutala, Weber and HICPAC 2002). For example, laparoscopes, which would be damaged if submitted to either high-pressure steam (autoclaving) or dry heat sterilization, usually are processed between cases by HLD (i.e., soaking in a chemical high-level disinfectant for 20 minutes). When correctly performed, sterilization clearly is the safest and

[4] Throughout this manual, when hepatitis B (HBV) is mentioned, hepatitis C (HCV) and Delta hepatitis (HDV) also are referred to because their occurrence is worldwide and mode of transmission or prevention is similar.

most effective method for the final processing of instruments. If it is neither available nor suitable, however, HLD is the **only** acceptable alternative for final processing.

High-level disinfection kills all microorganisms but does **not** reliably kill bacterial endospores. Staff must be aware of this limitation if tetanus, a disease caused by endospores produced by bacteria called *Clostridium tetani*, is a significant risk. The information in **Table 1-2** will assist healthcare providers and managers in determining when sterilization is preferable to HLD in processing surgical instruments and other reusable items. In addition, as a further guide, throughout this manual frequent reference is made to the limitations of HLD (i.e., does not reliably kill some endospores).

Table 1-2. Which Final Process to Use

PROCEDURE	STERILIZATION	HLD
Cesarean section	Preferred	Acceptable
Abdominal laparotomy	Preferred	Acceptable
Vaginal delivery	Preferred	Acceptable
Norplant implants insertion and removal	Preferred	Acceptable
Laparoscopy	Preferred (chemical only)	Acceptable
MVA cannulae[a]	Acceptable	Acceptable
IUD insertion and removal	Acceptable	Acceptable
Pelvic examination	Acceptable	Acceptable

[a] MVA: manual vacuum aspiration (for treatment of incomplete abortion)

Adapted from: Tietjen, Cronin and McIntosh 1992.

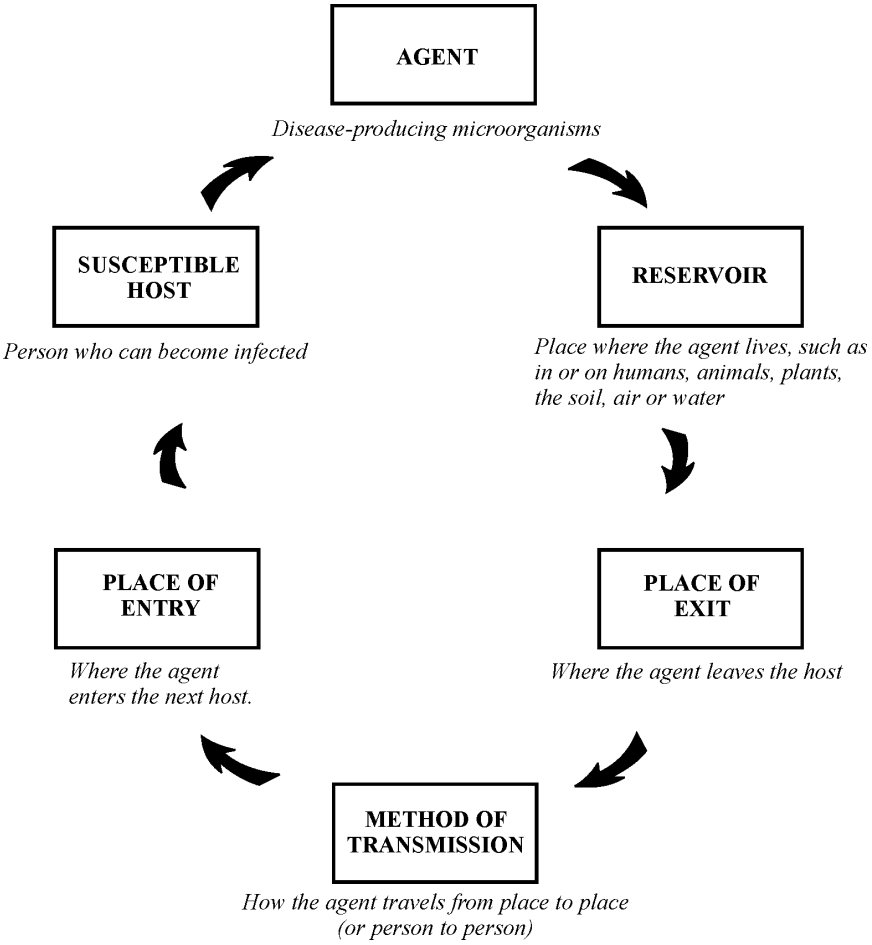
THE DISEASE TRANSMISSION CYCLE

Microorganisms live everywhere in our environment. Humans normally carry them on their skin and in the upper respiratory, intestinal and genital tracts. In addition, microorganisms live in animals, plants, soil, air and water. Some microorganisms, however, are more **pathogenic** than others, that is, they are more likely to cause disease. Given the right circumstances, **all** microorganisms may cause infection, such as when transmitted to an immunocompromised patient with AIDS (Burke 1977).

All humans are susceptible to bacterial infections and also to most viral agents. The dose of organisms (inoculum) necessary to produce infection in a susceptible host varies with the location. When organisms come in contact with bare skin, infection risk is quite low, and all of us touch materials that contain some organisms every day. When the organisms come in contact with mucous membranes or nonintact skin, infection risk increases. Infection risk increases greatly when organisms come in contact with normally sterile body sites, and the introduction of only a few organisms may produce disease.

For bacteria, viruses and other infectious agents to successfully survive and spread, certain factors or conditions must exist. The essential factors in the transmission of disease-producing microorganisms from person to person are illustrated and defined in **Figure 1-1** (APIC 1983; WPRO/WHO 1990).

Figure 1-1. The Disease Transmission Cycle



Adapted from: APIC 1983; WPRO/WHO 1990.

As shown in this figure, a disease needs certain conditions in order to spread (be transmitted) to others:

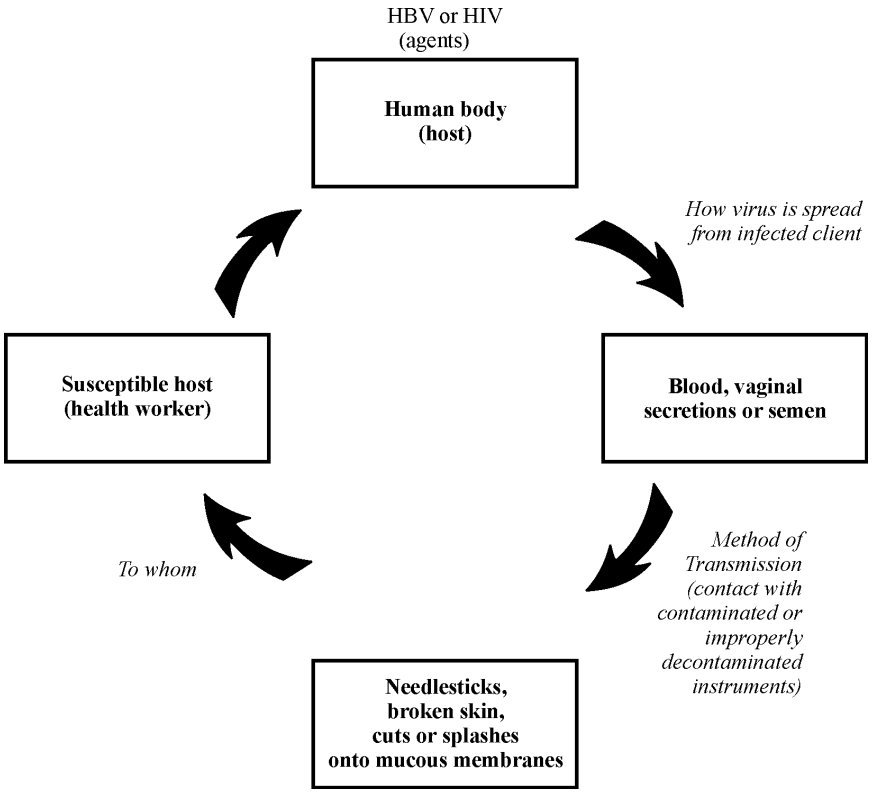
- There must be an **agent**—something that can cause illness (virus, bacteria, etc.).
- The agent must have a place it can live (**host** or **reservoir**). Many microorganisms that cause disease in humans (pathogenic organisms) multiply in humans and are transmitted from person to person. Some are transmitted through contaminated food or water (typhoid), fecal matter (hepatitis A and other enteric viruses) or the bites of infected animals (rabies) and insects (malaria from mosquitoes).
- The agent must have the right environment outside the host to survive. After the microorganism leaves its host, it must have a suitable environment in which to survive until it infects another person. For example, the bacteria that cause tuberculosis can survive in sputum for weeks, but will be killed by sunlight within a few hours.
- There must be a person who can catch the disease (**susceptible host**). People are exposed to disease-causing agents every day but do not always get sick. For a person to catch an infectious disease (e.g., mumps, measles or chicken pox,) s/he must be susceptible to that disease. The main reason most people do not catch the disease is that they have been previously exposed to it (e.g., vaccinated for it or previously had the disease) and their body's immune system now is able to destroy the agents when they enter the body.
- An agent must have a way to move from its host to infect the next susceptible host. Infectious (communicable) diseases are spread mainly in these ways:

    - **Airborne**: through the air (chicken pox or mumps).
    - **Blood or body fluids**: if blood or body fluids contaminated with HBV or HIV comes in contact with another person, such as through a needlestick, s/he may become infected.
    - **Contact**: either **direct** (touching an open wound or draining pustule), or **indirect** (touching an object contaminated with blood or other body fluids).
    - **Fecal-oral**: swallowing food contaminated by human or animal feces (e.g., putting your fingers in your mouth after handling contaminated objects without first washing your hands).
    - **Foodborne**: eating or drinking contaminated food or liquid that contains bacteria or viruses (hepatitis A from eating raw oysters).
    - **Animal- or insect-borne**: contact with infected animals or insects through bites, scratches, secretions or waste.

Infection prevention deals primarily with preventing the spread of infectious diseases through the air, blood or body fluids, and contact, including fecal-oral and foodborne.

Figure 1-2 depicts the steps in the transmission of the hepatitis B (HBV) and human immunodeficiency (HIV) viruses from colonized persons (e.g., family planning client or pregnant woman attending an antenatal clinic) or patients to healthcare workers. Spread of these viruses from person to person can occur when staff (physician, nurse or housekeeping personnel) are exposed to the blood or body fluids of an infected person (e.g., needlestick injury).

Figure 1-2. Transmission of HBV and HIV from Patients to Healthcare Workers



Studies in the United States have shown that the risk of disease after exposure to HBV from a single needlestick injury ranges from 27–37% (Seeff et al 1978), while the risk following a single needlestick exposure to HIV is much lower, 0.2–0.4% (Gerberding 1990; Gershon et al 1995), and 3–10% for HCV (Lanphear 1994). The rate of transmission of HIV is considerably lower than for HBV, probably because of the lower concentration of virus in the blood of HIV-infected persons.

The efficiency for transmission of hepatitis B is high. For example, an accidental splash in the eye of as little as 10^{-8} mL (.00000001 mL) of infected blood can transmit HBV to a susceptible host (Bond et al 1982).

In nearly all cases, transmission of HBV or HIV to health workers has occurred through preventable accidents such as puncture wounds. Transmission can also occur through mucous membrane contact, such as a

splash of blood or amniotic fluid into the surgeon's or assistant's eye. Also, skin damaged by a cut, scrape, chapped skin or contact dermatitis can be a point of entry for these viruses. While the risk of transmission is much lower from splashes of blood onto mucous membranes, they should be avoided. If splashing is anticipated, personal protective equipment such as face shields or glasses and plastic or rubber aprons, if available, is recommended. This protection is important because large mucous membrane exposures and prolonged skin contact may be associated with a higher risk of becoming infected (DHMH 1990).

Finally, because it is not always possible to know in advance whether or not a person may be infected with HBV or HIV, contaminated instruments, needles and syringes as well as other items from **all** persons (e.g., patients, pregnant women and other clients) must be handled as if they are contaminated. This practice is consistent with the recommendations in the new Standard Precaution Guidelines discussed in the next section (Garner and HICPAC 1996). For example, several studies have highlighted the inability to distinguish HBV- or HIV-infected people from noninfected individuals on clinical grounds (Baker et al 1987; Handsfield, Cummings and Swenson 1987; Kelen et al 1988).

PREVENTING INFECTIOUS DISEASES

Understanding the disease transmission cycle is important if healthcare workers are to:

- prevent transmission of microorganisms to patients during medical and surgical procedures;
- teach others the factors required for transmission to occur and, most importantly;
- teach others **how to** break the cycle.

Preventing the spread of infectious diseases requires removing one or more of the conditions necessary for transmission of the disease from host or reservoir to the next susceptible host by:

- inhibiting or killing the agent (e.g., applying an antiseptic agent to the skin before surgery);
- blocking the agent's means of getting from an infected person to a susceptible person (e.g., handwashing or using a waterless, alcohol-based antiseptic handrub to remove bacteria or viruses acquired through touching an infected patient or contaminated surface);
- making sure that people, especially healthcare workers, are immune or vaccinated; and

- providing health workers with the right protective equipment to prevent contact with infectious agents (e.g., heavy-duty gloves for housekeeping and waste removal staff).

NEW ISOLATION GUIDELINES AND RECOMMENDATIONS

Since 1970, when CDC first introduced the disease-specific category system of isolation precautions, many different policies and practices to prevent the spread of infections in hospitals have been recommended. Traditionally, barrier precautions (e.g., hand hygiene and gloves) have been used to reduce the risk of transmission of nosocomial infections to and from hospitalized patients. The emergence of bloodborne diseases such as AIDS and hepatitis C (HCV) in the 1980s, coupled with the resurgence of tuberculosis, first led to the introduction of Universal Precautions (UP) in 1985 and subsequently Body Substance Isolation (BSI) (1987). While many hospitals quickly began using some or all of the recommendations, there was much local variation and confusion in the use and interpretation of both UP and BSI. Thus, in 1996 the CDC and the Hospital Infection Control Practices Advisory Committee (HICPAC) issued a new system of isolation precautions (Garner and HICPAC 1996). This system involves a two-level approach—**Standard Precautions** and **Transmission-Based Precautions—**and was developed to meet the following criteria:

- Be epidemiologically sound
- Recognize the pathogenic importance of all body fluids, secretions and excretions (except sweat)
- Contain adequate precautions for infections transmitted by airborne, droplet or contact routes
- Be as simple and user-friendly as possible
- Use new terms to avoid confusion with existing systems

The new system accomplishes the following:

- Incorporates the major features of both UP and BSI into a single set of precautions, called **Standard Precautions**, that are designed to be used in treating **all clients** and **patients** attending healthcare facilities regardless of their presumed diagnosis.
- Retains the recommendations that healthcare workers providing direct care, especially those working in surgical or obstetrical units, should be immune to rubella, measles, mumps, varicella (chicken pox) and hepatitis A and B, as well as receive tetanus toxoid.

- Collapses the old disease-specific isolation categories into three sets of precautions based on routes of transmission, called **Transmission-Based Precautions**. (These guidelines apply to **hospitalized patients** or those in nursing homes or other types of extended care facilities.)
- Lists specific clinical syndromes in **hospitalized adult** and **child patients** that are highly suspicious for infection (i.e., the so called "empiric use" of Transmission-Based Precautions).

The new isolation guidelines are yet another positive step intended to reduce the risk of transmitting infections not only to and from patients and clients using healthcare services, but also to the healthcare personnel caring for them. As such, healthcare administrators and staff will need to carefully review the recommendations to determine what is possible, practical and doable within their resource setting.

Standard Precautions

Standard Precautions are designed for use in caring for **all people**—both clients and patients—attending healthcare facilities. They apply to blood, all body fluids, secretions and excretions (except sweat), nonintact skin and mucous membranes. Implementing these precautions, however, will add additional cost for personal protective equipment, especially for new examination gloves, staff training and monitoring in order to be effective. Because no one really knows what organisms clients or patients may have at any time, it is essential that Standard Precautions be used all the time. The details of their use and issues related to implementing them are covered in **Chapter 2**.

Transmission-Based Precautions

The second level of precautions is intended for use in patients **known** or **highly suspected** of being infected or colonized with pathogens transmitted by:

- air (tuberculosis, chicken pox, measles, etc.);
- droplet (flu, mumps and rubella); or
- contact (hepatitis A or E and other enteric pathogens, herpes simplex, and skin or eye infections).[5]

Note: In all cases, whether they are used alone or in combination, **Transmission-Based Precautions** must be used in conjunction with the **Standard Precautions**.

If there is any question of an infectious process in a patient without a known diagnosis, implementing Transmission-Based Precautions should be based on the patient's signs and symptoms (empiric basis) until a definitive diagnosis is made.

Use of Transmission-Based Precautions, including their empiric use, is designed to reduce the risk of spreading infections between hospitalized patients and healthcare staff. Occasionally, a patient may require isolation

[5] Contact precautions also should be used for patients with wet or draining infections that may be contagious (e.g., draining abscesses, herpes zoster, impetigo, conjunctivitis, scabies, lice and wound infections).

precautions involving more than one category. Their use is described in more detail in **Chapter 21**.

REFERENCES

Alvarado CJ. 2000. *The Science of Hand Hygiene: A Self-Study Monograph*. University of Wisconsin Medical School and Sci-Health Communications. March.

Association of Operating Room Nurses (AORN). 1990. Clinical issues. *AORN J* 52: 613–615.

Association for Practitioners in Infection Control (APIC). 1983. *The APIC Curriculum for Infection Control Practice,* Vol 1. Soule BM (ed). Kendall/Hunt Publishing Co.: Dubuque, IA, p 26.

Baker JL et al. 1987. Unsuspected human immunodeficiency virus in critically ill emergency patients. *JAMA* 257(19): 2609–2611.

Bond WW et al. 1982. Transmission of type B hepatitis via inoculation of a chimpanzee. *J Clin Microbiol* 15(3): 533–534.

Burke JF et al. 1977. The contribution of a bacterially isolated environment to the prevention of infection in seriously burned patients. *Ann Surg* 186(3): 377–387.

Department of Health and Mental Hygiene (DHMH). 1990. Occupational exposure to human immunodeficiency virus. *Communicable Disease Bulletin*. State of Maryland, December.

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80 and *Am J Infect Control* 24(1): 24–52.

Gerberding JL. 1990. Current epidemiologic evidence and case reports of occupationally acquired HIV and other bloodborne diseases. *Infect Control Hosp Epidemiol* 11(Suppl): 558–560.

Gershon RR et al. 1995. Compliance with universal precautions among health care workers at three regional hospitals. *Am J Infect Control* 23(4): 225–236.

Handsfield HH, MJ Cummings and PD Swenson. 1987. Prevalence of antibody to human immunodeficiency virus and hepatitis B surface antigen in blood samples submitted to a hospital laboratory: implications for handling specimens. *JAMA* 258(23): 3395–3397.

Jagger J et al. 1988. Rates of needlestick injury caused by various devices in a university hospital. *N Engl J Med* 319(5): 284–288.

Kelen GD et al. 1988. Unrecognized human immunodeficiency virus infection in emergency department patients. *N Engl J Med* 318(25): 1645–1650.

Lanphear B. 1994. Trends and patterns in the transmission of bloodborne pathogens to health care workers. *Epidemiol Rev* 16(2): 437–450.

Lynch P et al. 1997. *Infection Prevention with Limited Resources*. ETNA Communications: Chicago.

Nyström B. 1981. Disinfection of surgical instruments. *J Hosp Infect*. 2(4): 363–368.

Phipps W et al. 2002. Risk of medical sharps injuries among Chinese nurses. *Am J Infect Control* 30(5): 277–282.

Regional Office for the Western Pacific (WPRO), World Health Organization (WHO). 1990. *Infection Control*. HIV/AIDS Reference Library for Nurses.

Rogers B. 1997. Health hazards in nursing and health care: An overview. *Am J Infect Control* 25(3): 248–261.

Rutala WA, DJ Weber and The Hospital Infection Control Practices Advisory Committee (HICPAC). 2002. Draft guidelines for disinfection and sterilization in healthcare facilities. CDC corrected and cleared. February 2002.

Rutala WA. 1993. Disinfection, sterilization, and waste disposal, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RP (ed). Williams & Wilkins: Baltimore, MD, pp 460–495.

Seeff LB et al. 1978. Type B hepatitis after needlestick exposure: Prevention with hepatitis B immunoglobulin. Final report of the Veterans Administration Cooperative Study. *Ann Intern Med* 88(3): 285–293.

Spaulding EH. 1968. Chemical disinfection of medical and surgical materials, in *Disinfection, Sterilization and Preservation*. Lawrence CA et al (eds). Lea & Febiger: Philadelphia, pp 437–446.

Tietjen LG. 1997. Preventing infections in healthcare workers. *Outlook* 15(4): 1–4.

Tietjen LG, W Cronin and N McIntosh. 1992. Introduction to infection prevention, in *Infection Prevention Guidelines for Family Planning Service Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 1–12.

Tietjen LG and N McIntosh. 1989. Infection control in family planning facilities. *Outlook* 7(2): 2–8.

TWO

STANDARD PRECAUTIONS

KEY CONCEPTS you will learn in this chapter include:

- What the reasons for the new Standard Precautions are
- What Standard Precautions are designed to do
- What preventive processes and practices are recommended
- How protective barriers can help prevent the spread of infections

BACKGROUND

In 1985, largely because of the emergence of HIV/AIDS, guidelines for protecting healthcare workers from becoming infected with HIV and other bloodborne infections (e.g., HCV) were quickly developed and became known as Universal Precautions (UP). Almost from the moment they were issued and hospitals and clinics began implementing them, it was recognized that this new strategy, while protecting hospital personnel (patient-to-personnel transmission), sacrificed some measures of preventing patient-to-patient and personnel-to-patient transmission. Also, because many people with bloodborne infections such as HIV/AIDS do not have symptoms, nor can they be visibly recognized as being infected, UP had to be modified to include all persons—patients and clients—attending healthcare facilities regardless of whether or not they are infected (CDC 1985).

At nearly the same time that UP were being introduced, a new system of health worker and patient precautions was proposed as an alternative to the diagnosis-driven UP (Lynch et al 1987). This approach, called Body Substance Isolation (BSI), focused on protecting patients and health personnel from **all** moist and potentially infected body substances (secretions and excretions), not just blood. BSI was based primarily on the use of gloves. Personnel were instructed to put on clean gloves just before touching mucous membranes or nonintact skin, and before anticipated contact with moist body fluids (e.g., blood, semen, vaginal secretions, wound drainage, sputum, saliva, amniotic fluid, etc.). Other issues addressed by BSI included:

- protective immunization of susceptible patients and staff against infectious diseases that are transmitted by airborne or droplet routes (measles, mumps, chicken pox and rubella), as well as hepatitis A and B and tetanus toxoid immunization (or a booster dose) of staff; and

- revised instructions to persons wishing to enter a patient's room or care for patients with infections transmitted by the airborne route (Lynch et al 1990).

BSI quickly gained acceptance over UP because it was simple, easy to learn and implement, and acknowledged that all patients, not just those diagnosed or with symptoms, may be infected and therefore not free of risk to other patients or staff. Disadvantages of BSI included the added cost of protective barrier equipment, particularly gloves, difficulty in maintaining routine use of the protocol for all patients, uncertainty about precautions for patients in isolation rooms and the overuse of gloves to protect staff at the expense of patients (Patterson et al 1991).

As a consequence, by the early 1990s healthcare facilities and staff were totally confused regarding what to do about patient and staff precaution guidelines. For example, some hospitals had implemented UP while others had implemented BSI. Indeed, even hospitals and staff that thought they were following UP were really using BSI, and vice versa. There was also much local variation in interpretation and use of both UP and BSI, and a variety of combinations was common. Moreover, there was continued lack of agreement about the role of handwashing when gloves were used. This confusion, coupled with the need to use additional precautions to prevent diseases spread by airborne, droplet and contact routes, were major limitations of BSI (Rudnick et al 1993).

In view of these problems and concerns, no simple merging together of UP or BSI appeared likely to solve them. What has emerged since then is a new system that provides a single set of isolation guidelines with logistically feasible recommendations for preventing the many infections that occur in healthcare facilities through all known modes of transmission.

STANDARD PRECAUTIONS

The new guidelines issued by CDC in 1996 involve a two-level approach:

- **Standard Precautions,** which apply to **all clients and patients** attending healthcare facilities, and
- Transmission-Based Precautions, which apply only to hospitalized patients (Garner and HICPAC 1996).

As briefly presented in **Chapter 1**, this new system retains features of both UP and BSI. Moreover, it replaces the cumbersome disease-specific isolation precautions with three sets of transmission-based precautions for use in hospitalized patients.

Because most people with bloodborne viral infections such as HIV and HBV do not have symptoms, nor can they be visibly recognized as being infected, Standard Precautions are designed for the care of **all** persons—patients, clients and staff—regardless of whether or not they are infected. Standard Precautions apply to blood and all other body fluids, secretions and excretions (except sweat), nonintact skin and mucous membranes. Their implementation is meant to reduce the risk of transmitting microorganisms from known or unknown sources of infection (e.g., patients, contaminated objects, used needles and syringes, etc.) within the healthcare system. Applying Standard Precautions has become the primary strategy to preventing nosocomial infections in hospitalized patients.

Over the years, the indications for use of certain isolation practices over others (e.g., clean gloves are more effective than gowns in preventing cross-contamination) have been largely resolved through research (LeClaire et al 1987). However, the inability of hospital and clinic administrators in resource-poor countries to provide the required protective equipment, especially sufficient new examination gloves, remains a problem. In addition, the challenges of providing clean water and achieving acceptable standards of medical instrument processing and waste removal remain unmet in many countries. In most cases, staff training to implement these new isolation precautions for every client attending a clinic or every hospitalized patient will require that resources be shifted from one priority area to another. Moreover, the regular supervision needed to assure compliance is seldom affordable or available. As a consequence, healthcare administrators and staff will need to carefully review the recommendations contained in the Standard Precautions and modify them according to what is possible and practical within their resource setting.

KEY COMPONENTS AND THEIR USE

The key components of the Standard Precautions and their use are outlined in **Table 2-1**. Placing a physical, mechanical or chemical **barrier** between microorganisms and an individual—whether a woman coming for antenatal care, a hospitalized patient or healthcare worker—is a highly effective means of preventing the spread of infections (i.e., the barrier serves to break the disease transmission cycle). For example, the following actions create protective barriers for preventing infections in clients, patients and healthcare workers and provide the means for implementing the new Standard Precautions:

- **Consider every person** (patient or staff) as potentially infectious and susceptible to infection.
- **Wash hands—**the most important procedure for preventing cross-contamination (person to person or contaminated object to person).

- **Wear gloves** (both hands) before touching anything wet—broken skin, mucous membranes, blood or other body fluids, or soiled instruments and contaminated waste materials—or before performing invasive procedures.

Table 2-1. Standard Precautions: Key Components

Handwashing (or using an antiseptic handrub)

- After touching blood, body fluids, secretions, excretions and contaminated items
- Immediately after removing gloves
- Between patient contact

Gloves

- For contact with blood, body fluids, secretions and contaminated items
- For contact with mucous membranes and nonintact skin

Masks, goggles, face masks

- Protect mucous membranes of eyes, nose and mouth when contact with blood and body fluids is likely

Gowns

- Protect skin from blood or body fluid contact
- Prevent soiling of clothing during procedures that may involve contact with blood or body fluids

Linen

- Handle soiled linen to prevent touching skin or mucous membranes
- Do not pre-rinse soiled linens in patient care areas

Patient care equipment

- Handle soiled equipment in a manner to prevent contact with skin or mucous membranes and to prevent contamination of clothing or the environment
- Clean reusable equipment prior to reuse

Environmental cleaning

- Routinely care, clean and disinfect equipment and furnishings in patient care areas

Sharps

- Avoid recapping used needles
- Avoid removing used needles from disposable syringes
- Avoid bending, breaking or manipulating used needles by hand
- Place used sharps in puncture-resistant containers

Patient resuscitation

- Use mouthpieces, resuscitation bags or other ventilation devices to avoid mouth-to-mouth resuscitation

Patient placement

- Place patients who contaminate the environment or cannot maintain appropriate hygiene in private rooms

- **Use physical barriers** (protective goggles, face masks and aprons) if splashes and spills of any body fluids (secretions and excretions) are likely (e.g., cleaning instruments and other items).

- **Use antiseptic agents** for cleansing the skin or mucous membrane prior to surgery, cleaning wounds, or doing handrubs or surgical handscrubs with an alcohol-based antiseptic product.
- **Use safe work practices** such as not recapping or bending needles, safely passing sharp instruments and suturing, when appropriate, with blunt needles.
- **Safely dispose of infectious waste materials** to protect those who handle them and prevent injury or spread of infection to the community.
- **Process instruments, gloves and other items** after use by first decontaminating and thoroughly cleaning them, then either sterilizing or high-level disinfecting them using the recommended procedures.

The use of these precautions, including hand hygiene and prevention-related services such as immunization of healthcare workers, is fully described in **Chapters 3–8** and **Appendices A–D**. Details of how to safely process soiled instruments, gloves and other items are presented in **Chapters 9–14** and **Appendices E–H**.

REFERENCES

Centers for Disease Control (CDC). 1985. Recommendations for preventing transmission of infection with human T-lymphotropic virus type III/lymphadenopathy-associated virus in the workplace. *MMWR* 34(45): 681–686; 691–695.

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80 and *Am J Infect Control* 24(1): 24–52.

Lynch P et al. 1990. Implementing and evaluating a system of generic infection precautions: Body substance isolation. *Am J Infect Control* 18(1): 1–12.

Lynch P et al. 1987. Rethinking the role of isolation practices in the prevention of nosocomial infections. *Ann Intern Med* 107(2): 243–246.

LeClaire JM et al. 1987. Prevention of nosocomial respiratory syncytial virus infections through compliance with glove and gown isolation precautions. *N Engl J Med* 317(6): 329–334.

Patterson JE et al. 1991. Association of contaminated gloves with transmission of *Acinetobacter calcoaceticus var. anitratus* in an intensive care unit. *Am J Med* 91(5): 479–483.

Rudnick JR et al. 1993. Are US hospitals prepared to control nosocomial transmission of tuberculosis? Epidemic Intelligence Service Annual Conference 60: Abstract.

THREE

HAND HYGIENE

KEY CONCEPTS you will learn in this chapter include:

- Why hand hygiene is important
- When and how to wash hands
- When and how to use waterless, alcohol-based antiseptic agents for handrub and surgical handscrub
- What the barriers to appropriate hand hygiene are
- How to improve hand hygiene practices in healthcare facilities

BACKGROUND

Failure to perform appropriate hand hygiene is considered to be the leading cause of nosocomial (hospital-acquired) infections and the spread of multiresistant microorganisms, and has been recognized as a significant contributor to outbreaks (Boyce and Pittet 2002).

Hand hygiene guidelines, which traditionally have dealt with recommendations for when and how to perform handwashing or surgical handscrubs, have undergone rapid change in the past 15 years. With the emergence of the AIDS epidemic in the late 1980s, efforts to prevent transmission of HIV and other bloodborne viruses from patients to staff have had an impact on all aspects of infection prevention, but most dramatically on hand hygiene and glove use practices. For example, current hand hygiene guidelines must not only provide solid evidence in support of recommendations to promote improved practices, but increasingly they must also deal with issues of:

- poor compliance;
- how to minimize skin irritation and contact dermatitis resulting from frequent handwashing; and
- use of waterless, alcohol-based antiseptic handrubs, as well as moisturizing lotions and creams by healthcare personnel.

The criteria for handwashing or use of an antiseptic handrub are presented in **Table 3-1**.

Table 3-1. Criteria for Handwashing or Use of an Antiseptic Handrub

The decision to clean hands is based on:

- intensity of contact with patient and/or blood and body fluids,
- likelihood of microbial transmission,
- patient's susceptibility to infection, and
- procedure being performed.

The use of soap and water remains important when hands are visibly soiled. For routine hand hygiene in the absence of dirt or debris, however, alternatives such as antiseptic handrubs, which are rapid acting, inexpensive and easy to make, are gaining acceptance, especially where access to sinks and clean water is limited.[1]

From the point of view of infection prevention, hand hygiene practices (handwashing and surgical handscrubbing) are intended to prevent hand-borne infections by removing dirt and debris and inhibiting or killing microorganisms on skin. This includes not only most of the organisms acquired through contact with patients and the environment, but also some of the permanent ones that live in the deeper layers of the skin. In addition to understanding the guidelines and recommendations for hand hygiene, healthcare workers need to understand the value, and especially the limitations, of glove use (see **Chapter 4**).

A key first step in this process is educating health professional students and healthcare workers about:

- the importance of hand hygiene, how to correctly perform the various handwashing and handscrubbing procedures; and
- the evidence supporting the use of these procedures in reducing transmission of microorganisms and therefore decreasing the frequency of nosocomial infections in patients.

Finally, not only can frequent handwashing reduce the spread of infection from the hands of health workers, but from everyone else's as well! For example, it is estimated that persuading people, especially young children, to wash their hands with soap and clean water after going to the toilet, handling or changing a dirty baby, or doing other tasks that potentially contaminate hands (cleaning vegetables, fresh meat or fish) can reduce diarrheal diseases by 45%, saving the lives of a million children a year (*The Economist* 2002). Moreover, in a large study, the US military found that when troops washed

[1] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

their hands five or more times daily, sniffles, coughs and common "colds" fell by 43%.

DEFINITIONS

- **Antiseptic** or **antimicrobial agent (terms used interchangeably)**. Chemicals that are applied to the skin or other living tissue to inhibit or kill microorganisms (both transient and resident) thereby reducing the total bacterial counts. Examples include alcohols (ethyl and isopropyl), dilute iodine solutions, iodophors, chlorhexidine and triclosan. (See **Appendix B** for complete listing of uses, effectiveness, advantages and disadvantages of selected antiseptic agents.)
- **Clean water**. Natural or chemically treated and filtered water that is safe to drink and use for other purposes (e.g., handwashing and medical instrument cleaning) because it meets specified public health standards. These standards include: zero levels of microorganisms, such as bacteria (e.g., fecal coliform and *Escherichia coli*), parasites (e.g., *Giardia lamblia*) and viruses (e.g., hepatitis A or E); low turbidity (cloudiness due to particulate matter and other contaminants); and minimum levels of disinfectants, disinfectant by-products, inorganic and organic chemicals and radioactive materials. At a minimum clean water should be free of microorganisms and have low turbidity (is clear, not cloudy).
- **Emollient**. Organic liquid, such as glycerol, propylene glycol or sorbitol, that when added to handrubs and hand lotions softens the skin and helps prevent skin damage (cracking, drying, irritation and dermatitis) due to frequent handwashing with soap (with or without antiseptic agent) and water.
- **Handwashing**. Process of mechanically removing soil and debris from the skin of hands using plain soap and water.
- **Nosocomial** or **hospital-acquired infection (terms used interchangeably)**. Infection that is neither present nor incubating at the time the patient came to the healthcare facility. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.)
- **Soaps** and **detergents (terms used interchangeably)**. Cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands. **Plain** soaps require friction (scrubbing) to mechanically remove microorganisms, while **antiseptic** (antimicrobial) soaps also kill or inhibit growth of most microorganisms.
- **Transient** and **resident flora**. Terms that refer to where bacteria and other microorganisms are located in the layers of the skin. **Transient flora** are acquired through contact with patients, other healthcare workers or contaminated surfaces (e.g., examination tables, floors or toilets) during the course of the normal workday. These organisms live in the

upper layers of the skin and are partially removed by washing with plain soap and clean water. They are the organisms most likely to cause nosocomial infections. **Resident flora** live in the deeper layers of the skin, as well as within hair follicles, and cannot be completely removed, even by vigorous washing and rinsing with plain soap and clean water. Fortunately, in most cases, resident flora is less likely to be associated with infections. The hands or fingernails of some health workers, however, can become colonized in the deep layers with organisms that cause infections, such as *S. aureus,* gram-negative bacilli or yeast.

- **Visibly soiled hands**. Hands showing visible dirt or are visibly contaminated with blood or body fluids (urine, feces, sputum or vomit).
- **Waterless, alcohol-based antiseptic handrub** or **antiseptic handrub (terms used interchangeably)**. Fast acting antiseptic handrubs that do not require use of water to remove transient flora, reduce resident microorganisms and protect the skin. Most contain 60–90% alcohol, an emollient and often an additional antiseptic (e.g., 2–4% chlorhexidine gluconate) that has residual action (Larson et al 2001).

HAND HYGIENE PRACTICES

Hand hygiene significantly reduces the number of disease-causing microorganisms on hands and arms and can minimize cross-contamination (e.g., from health worker to patient). The indications for hand hygiene are well known, but guidelines for best practices continue to evolve. For example, the choice of plain or antiseptic soap, or use of an antiseptic handrub, will depend on the degree of risk with patient contact (e.g., routine medical procedure versus surgery) and availability (Larson 1995). Current recommendations for healthcare workers are:

- When skin is damaged or frequent handwashing is required, a mild soap (without antiseptic agent) should be used to remove soil and debris.
- If antimicrobial action is desired (e.g., before an invasive procedure or contact with highly susceptible patients such those with AIDS or newborns) and hands are not visibly dirty, an antiseptic handrub should be used rather than washing hands with medicated antiseptic soap.
- In high-risk areas such as the operating room, neonatal ICU or transplant units, handscrub protocols that use soft brushes or sponges for a shorter time (at least 2 minutes) should replace harsh scrubbing with hard brushes for 6–10 minutes.
- For staff who frequently wash their hands (30 times or more per shift), hand lotions and creams should be provided in order to reduce irritation of the skin.

Hand hygiene can be accomplished by routine handwashing (with or without antiseptic agent) or antiseptic handrub and surgical handscrub using a

waterless, alcohol-based antiseptic agent. The purpose and way to do each differs slightly.

Handwashing

The purpose of handwashing is to mechanically remove soil and debris from the skin and reduce the number of transient microorganisms. Handwashing with plain soap and **clean** water is as effective as washing with antimicrobial soaps (Pereira, Lee and Wade 1997).[2] In addition, plain soap causes much less skin irritation (Pereira, Lee and Wade 1990).

Handwashing should be done **before**:

- examining (direct contact with) a patient; and
- putting on **sterile** or **high-level disinfected** surgical gloves prior to an operation, or examination gloves for routine procedures such as a pelvic examination.

Handwashing should be done **after**:

- any situation in which hands may become contaminated, such as:
  - handling soiled instruments and other items;
  - touching mucous membranes, blood or other body fluids (secretions or excretions);
  - having prolonged and intense contact with a patient; and
- removing gloves.

Hands should be washed with soap and clean water (or an antiseptic handrub can be used) **after** removing gloves because the gloves now may have tiny holes or tears, and bacteria can rapidly multiply on gloved hands due to the moist, warm environment within the glove (CDC 1989; Korniewicz et al 1990).

Note: If paper towels are not available, dry hands with a clean towel or air dry. (Shared towels quickly become contaminated and should not be used. Carrying one's own small towel or handkerchief can help to avoid using dirty towels. If you use your own towel, it should be washed every day.)

To encourage handwashing, program managers should make every effort to provide soap and a continuous supply of clean water, either from the tap or a bucket, and single-use towels.

The steps for routine handwashing are:

STEP 1: Thoroughly wet hands.

STEP 2: Apply plain soap (antiseptic agent is not necessary).

STEP 3: Vigorously rub all areas of hands and fingers together for at least 10 to 15 seconds, paying close attention to areas under fingernails and between fingers.

[2] If tap water is contaminated, however, handwashing with plain soap is only effective in removing dirt and debris.

STEP 4: Rinse hands thoroughly with clean water.

STEP 5: Dry hands with a paper towel and use the towel to turn off the faucet.

Because microorganisms grow and multiply in moisture and in standing water:

Note: When soap dispensers are reused, they should be thoroughly cleaned before filling.

- If bar soap is used, provide small bars and soap racks that drain.
- Avoid dipping hands into basins containing standing water. Even with the addition of an antiseptic agent, such as Dettol® or Savlon®, microorganisms can survive and multiply in these solutions (Rutala 1996).
- Do not add soap to a partially empty liquid soap dispenser. This practice of "topping off" dispensers may lead to bacterial contamination of the soap.

Note: Used water should be collected in a basin and discarded in a latrine if a drain is not available.

- When no running water is available, use a bucket with a tap that can be turned off to lather hands and turned on again for rinsing, or use a bucket and pitcher.

Hand Antisepsis

The goal of hand antisepsis it to remove soil and debris as well as to reduce **both** transient and resident flora. The technique for hand antisepsis is similar to that for plain handwashing. It consists of washing hands with water and soap or detergent (bar or liquid) containing an antiseptic agent (often chlorhexidine, iodophors or triclosan) instead of plain soap.

Hand antisepsis should be done **before**:

- examining or caring for highly susceptible patients (e.g., premature infants, elderly patients or those with advanced AIDS);
- performing an invasive procedure such as placement of an intravascular device; and
- leaving the room of patients on Contact Precautions (e.g., hepatitis A or E) or who have drug resistant infections (e.g., methicillin-resistant *S aureus)*.

Handwashing with medicated soaps or detergents is more irritating to the skin than using antiseptic handrubs (see next section); therefore, if available, antiseptic handrubs should be used instead (Larson et al 1990 and Larson et al 2001).

Antiseptic Handrub

Use of an antiseptic handrub is more effective in killing transient and resident flora than handwashing with antimicrobial agents or plain soap and water, is quick and convenient to perform, and gives a greater initial reduction in hand flora (Girou et al 2002). Antiseptic handrubs also contain a small amount of

an emollient such as glycerin, propylene glycol or sorbitol that protects and softens skin.

The technique for performing antiseptic handrub is:

STEP 1: Apply enough antiseptic handrub to cover the entire surface of hands and fingers (about a teaspoonful).

STEP 2: Rub the solution vigorously into hands, especially between fingers and under nails, until dry.

To be effective, an adequate amount of handrub solution should be used. For example, by increasing the amount of handrub from 1 mL to 5 mL per application (about 1 teaspoonful), the effectiveness increased significantly (Larson 1988).

Since antiseptic handrubs do not remove soil or organic matter, if hands are visibly soiled or contaminated with blood or body fluids, handwashing with soap and water should be done first. In addition, to reduce the "build up" of emollients on hands after repeated use of antiseptic handrubs, washing hands with soap and water after every 5–10 applications is recommended. Finally, handrubs containing only alcohol as the active ingredient have limited residual effect (i.e., ability to prevent growth of bacteria after being applied) compared to those containing alcohol plus an antiseptic such as chlorhexidine.

As shown below, an effective antiseptic handrub solution is inexpensive and simple to make.

Alcohol-Based Solution for Handrub

A nonirritating, antiseptic handrub can be made by adding either glycerin[a], proplyene glycol or sorbitol to alcohol (2 mL in 100 mL of 60–90% ethyl or isopropyl alcohol solution) (Larson 1990; Pierce 1990). Use 5 mL (about one teaspoonful) for each application and continue rubbing the solution over the hands until they are dry (15–30 seconds).

[a] Glycerin is often sold in cosmetic departments because it is used as a hand softener.

Surgical Handscrub

The purpose of the surgical handscrub is to mechanically remove soil, debris and transient organisms and to reduce resident flora for the duration of surgery. The goal is to prevent wound contamination by microorganisms from the hands and arms of the surgeon and assistants.

For many years, preoperative handscrubbing protocols required at least a 6–10 minute vigorous scrub with a brush or sponge, using soap containing an antiseptic agent (chorhexidine or an iodophor). This practice, however, has been shown to damage the skin and can result in increased shedding of bacteria from the hands (Dineen 1966; Kikuchi-Numagami et al 1999). Several studies suggest that neither a brush nor sponge is necessary to reduce bacterial counts on the hands of surgical staff to acceptable levels. For example, a 2-minute handwashing with soap and clean water followed by application of 2–4% chlorhexidine or 7.5–10% povidone iodine was shown to be as effective as a 5-minute handscrub with an antiseptic soap (Deshmukh, Kramer and Kjellberg 1996; Pereira, Lee and Wade 1997). As a result, the guidelines for performing the general surgical scrub technique have been made less harsh and take less time to perform. The steps include:

STEP 1: Remove rings, watches and bracelets.

STEP 2: Thoroughly wash hands, especially between fingers, and forearms to the elbows with soap and water. (If a brush is used it should be cleaned and either sterilized or high-level disinfected before reuse; sponges, if used, should be discarded.)

STEP 3: Clean nails with a nail cleaner.

STEP 4: Rinse hands and forearms with water.

STEP 5: Apply an antiseptic agent to all surfaces of hands and forearms to the elbows and rub hands and forearms vigorously for at least 2 minutes.

STEP 6: Holding the hands higher than the elbows, rinse hands and forearms thoroughly with clean water.[3]

STEP 7: Keep hands up and away from the body, do not touch any surface or article and dry hands and forearms with a clean, dry towel or air dry.

STEP 8: Put sterile or high-level disinfected surgical gloves on both hands.

Applying an antiseptic minimizes the number of microorganisms on hands under the gloves and minimizes growth of flora during surgery. This is important because gloves may have inapparent holes or tears, or may be nicked during surgery.

(Complete instructions for how to do a general surgical handscrub are outlined in **Appendix A**.)

Note: Skin damage caused by allergic reactions provides an ideal place for microorganisms to multiply and should be avoided. (Personnel with allergies to antiseptics may use plain soap followed by applying the waterless, alcohol-based handrub described above.)

[3] If tap water is contaminated, use boiled or chlorinated water and filter if necessary.

Alternatively, handwashing with plain soap and water followed by use of an antiseptic handrub containing chlorhexidine has been shown to yield significantly greater reductions in microbial counts on hands, improve skin health and reduce time and resources (Larson et al 2001).

The steps for performing this simpler and shorter surgical handscrub technique are:

STEP 1: Remove rings, watches and bracelets.

STEP 2: Thoroughly wash hands, especially between fingers, and forearms to the elbows with soap and water.

STEP 3: Clean nails with a nail cleaner.

STEP 4: Rinse hands and forearms with water and dry thoroughly with a clean, dry towel or air dry.

STEP 5: Apply 5 mL (about 1 teaspoonful) of an antiseptic handrub to hands and forearms and rub until dry; repeat application and rubbing 2 more times for a total of at least 2 minutes, using a total of about 15 mL (3 teaspoonfuls) of the handrub.

STEP 6: Keep hands up and away from the body; do not touch any surface or article prior to putting sterile or high-level disinfected surgical gloves on both hands.

IMPROVING HAND HYGIENE PRACTICE: WHAT WORKS

Handwashing has been considered one of the most important measures for reducing transmission of microorganisms and preventing infection for more than 150 years. For example, the studies of Semmelweiss (1861) and numerous others since then have demonstrated it is possible to transmit infectious diseases from patient to patient on the hands of healthcare workers. Equally well documented is the fact that good hand hygiene can prevent transmission of microorganisms and decrease the frequency of nosocomial infections (Boyce 1999; Larson 1995).

The continuing problem, however, is getting healthcare workers to follow recommended handwashing practices. For example, in the US, handwashing compliance rates among healthcare workers range from only 25% to 50%, depending on the setting (i.e., better compliance in pediatric units than general medical services). Key reasons given for not washing hands according to recommended guidelines include lack of time, limited access to sinks and running water, frequent handwashing irritates the hands, belief that wearing gloves provides total protection, doubt regarding the effectiveness of handwashing to prevent infections, and perception that peers and supervisors do not perform handwashing as recommended (**Table 3-2**). In addition, health professionals mistakenly believe they wash their hands more often than they actually do (Tibballs 1996)!

Over the years, nurses and physicians have diligently studied and written about this problem. Numerous reports have documented the effectiveness of handwashing and other hand hygiene procedures and shown that handwashing and use of gloves are cost-effective ways to reduce infections. Despite this, compliance remains poor and the problem of nosocomial infections transmitted by healthcare workers continues to increase globally. To correct this situation, in the last few years several strategies to improve compliance have been designed and tested. Those showing the most promise combine behavior change activities, such as continuing education, motivation and system change, with role modeling or mentoring, and ongoing feedback to staff. While the results to date have not been totally successful, improvements have been demonstrated (i.e., reduced rates of nosocomial infections) in several studies (Larson et al 2000; Pittet et al 2000). In the future, other innovative approaches, such as education of patients and their families about the importance of staff handwashing, may also prove successful.

Table 3-2. Why Healthcare Professionals Don't Wash Their Hands

Belief that:

- Handwashing between every patient encounter is unnecessary
- Handwashing does not affect clinical outcome
- Handwashing is unnecessary when gloves are worn
- Routine or frequent handwashing is unnecessary
- Frequent handwashing interrupts efficient patient care
- Frequent handwashing damages skin and causes cracking, dryness, irritation and dermatitis
- Handwashing damages nails and nail polish
- Handwashing facilities are not conveniently placed or well designed
- Handwashing is inconvenient
- Handwashing takes too much time

Failure of supervisors and managers to:

- Establish a handwashing policy
- Involve administrators in handwashing policy
- Effectively communicate handwashing policy
- Demonstrate handwashing policy through actions
- Enforce handwashing policy

Adapted from: Alvarado 2000.

Although it is difficult to change behavior in this area, there are certain steps that increase the chances of success. These include:

- Widely disseminate current guidelines for hand hygiene practices, the evidence supporting their effectiveness in preventing disease and the need for health workers to adhere to the guidelines.
- Involve hospital administrators in promoting and enforcing the guidelines by convincing them of the cost benefits of handwashing and other hand hygiene practices.
- Use successful educational techniques including role modeling (especially by supervisors), mentoring, monitoring and positive feedback.
- Use performance improvement approaches targeted to all healthcare staff, not just physicians and nurses, to promote compliance.
- Consider the needs of staff for convenient and effective options for hand hygiene that make compliance easier.

One promising example of how to make compliance easier is providing staff with small, individual-use containers of an antiseptic handrub. Development of this product stems from the observation that improper handwashing techniques and low compliance make current hand hygiene recommendations ineffective. Use of an inexpensive, simple to prepare antiseptic handrub, however, minimizes many of the factors limiting better use of recommended hand hygiene guidelines. In addition, handrubs are more effective compared to washing hands with plain or medicated soaps, can be made much more available (no sink or running water needed), require less time to use and are less likely to irritate the skin (less drying, cracking or chapping). As a consequence, antiseptic handrubs soon may replace handwashing with plain or medicated soap and water as the primary procedure for improving compliance (Larson et al 2000; Pittet et al 2000). Interestingly, the only large-scale, hospital-based programs that reported sustained improvement in hand hygiene adherence associated with reduced infection rates incorporated use of antiseptic handrubs (Larson et al 2000; Pittet et al 2000). It must be recognized, however, that making a handrub available to staff without ongoing educational and motivational activities may not result in long-lasting improvement in hand hygiene practices. Just installing dispensers of a rapid acting, antiseptic handrub, for example, is not sufficient (Muto et al 2000).

A second example is encouraging staff to use handcare products (moisturizing lotions and creams) that help prevent skin irritation and contact dermatitis associated with frequent handwashing, especially with a soap or detergent containing an antiseptic agent. Not only were staff highly satisfied with the results but, most importantly, in the study by McCormick et al (2000), improved skin condition resulting from use of a hand lotion led to a 50% increase in handwashing frequency!

A final example, which illustrates the role teachers and supervisors can play in improving hand hygiene practices, relates to current guidelines still calling for handwashing **before** and **after** each patient contact. This recommendation is confusing because it does not take into account that washing **after** might be adequate if no hand contamination occurs before touching the next patient. Recognizing confusing hand hygiene guidelines and advocating for their improvement is also a way for teachers and supervisors to demonstrate their commitment. This also helps health workers to meet criteria both for using good hand hygiene and providing appropriate patient care.

In summary, although improving compliance with hand hygiene guidelines has been difficult, some programs and institutions are beginning to have success. The key to success appears to hinge on multifactored interventions that involve behavior change, creative education, monitoring and feedback and, above all, involvement of their supervisors as role models and the support of administration.

OTHER ISSUES AND CONSIDERATIONS RELATED TO HAND HYGIENE

Glove Use

Since 1987 and the emergence of the AIDS epidemic, a dramatic increase in glove use by all types of healthcare staff has occurred in an effort to prevent transmission of HIV and other bloodborne viruses from patients to staff. Although the effectiveness of gloves in preventing contamination of health workers' hands has been repeatedly confirmed (Tenorio et al 2001), preventing gross contamination of hands is considered important. For example, handwashing, even with an antiseptic agent, may not remove all potential pathogens when hands are heavily contaminated. These findings have mistakenly led some health workers to doubt the efficacy of hand hygiene practices under any circumstances, resulting in poor or infrequent use of handwashing by these staff.

Gloves, however, do not provide complete protection against hand contamination. For example, bacteria from patients can be recovered in up to 30% of staff who wear gloves during patient care (Kotilainen et al 1989). Also, oral surgeons wearing gloves and other protective devices have become infected with hepatitis B, presumably via small defects in the gloves or their hands becoming contaminated during glove removal (Reingold, Kane and Hightower 1988). Moreover, wearing the same pair of gloves and washing gloved hands between patients or between dirty to clean body site care is not a safe practice. Doebbeling and colleagues (1988) recovered significant amounts of bacteria on the hands of staff not changing gloves between patients, but just washing their gloved hands.

The overall influence of glove use on hand hygiene practices of staff is not clear, however. For example, some studies have reported that staff who wear gloves were less likely to wash their hands, while others have found the opposite. Given the generally poor compliance with hand hygiene practices,

every effort must be made to reinforce the message that gloves do not replace the use of hand hygiene, but in certain circumstances, gloves should be used in addition to hand hygiene.

Hand Lotions and Hand Creams

In an effort to minimize hand hygiene-related contact dermatitis due to frequent handwashing (>30 times per shift), use of harsh detergents and exposure to antiseptic agents (60–90% alcohol is less irritating to skin than any other antiseptic or nonantiseptic detergent), health workers have resorted to using hand lotions, creams and moisturizing skin care products. Several studies have shown that regular use (at least twice per day) of such products can help prevent and treat contact dermatitis (McCormick et al 2000). In addition, moisturizers can prevent drying and damage to the skin and loss of skin fats. There is also biological evidence that emollients, such as glycerol and sorbitol, with or without antiseptics, may decrease cross contamination because they reduce shedding of bacteria from skin for up to 4 hours.

While use of hand lotions, creams and moisturizers by health workers should be encouraged, it is recommended that the products be supplied in either small, individual-use containers that can be easily carried or in pump dispensers that cannot be refilled to reduce the possibility of becoming contaminated. (To avoid confusion, these dispensers should not be located near dispensers of antiseptic solutions.) By contrast, oil-based barrier products, such as those containing petroleum jelly (Vaseline® or lanolin), should not be used because they damage latex rubber gloves.

Resistance to Topical Antiseptic Agents

With increasing use of topical antiseptics, particularly in home settings, concern has been raised regarding the development of resistance. Although low-level bacterial tolerance to triclosan, a commonly used antiseptic agent, has been noted in several laboratory-based studies, prolonged clinical studies have found that extended use of triclosan-containing products does not lead to resistance of skin flora. Moreover, other studies have noted no clinical evidence to date that supports development of resistant organisms following use of any topical antiseptics agents.

Lesions and Skin Breaks

Cuticles, hands and forearms should be free of lesions (dermatitis or eczema) and skin breaks (cuts, abrasions and cracking). Cuts and abrasions should be covered with waterproof dressings. If covering them in this way is not possible, surgical staff with skin lesions should not operate until the lesions are healed.

Fingernails

Research has shown that the area around the base of nails (subungual space) contains the highest microbial count on the hand (McGinley, Larson and Leydon 1988). In addition, several recent studies have shown that long nails may serve as a reservoir for gram-negative bacilli (*P. aeruginosa*), yeast and other pathogens (Hedderwick et al 2000). Moreover, long nails, either natural or artificial, tend to puncture gloves more easily (Olsen et al 1993). As a result, it is recommended that nails be kept moderately short—not extend more than 3 mm (or 1/8 inch) beyond the fingertip.

Artificial Nails

Artificial nails (nail wraps, nail tips, acrylic lengtheners, etc.) worn by healthcare workers can contribute to nosocomial infections (Hedderwick et al 2000). In addition, because there is evidence that artificial nails may serve as a reservoir for pathogenic gram-negative bacilli, their use by health workers should be restricted, especially by surgical team members, and those who:

- work in specialty areas such as neonatal ICUs,
- care for patients highly susceptible to infection, or
- manage patients who have infections with resistant organisms (Moolenaar et al 2000).

Nail Polish

Although there is no restriction to wearing nail polish, it is suggested that surgical team members and those staff working in specialty areas wear freshly applied, clear nail polish. Chipped nail polish supports the growth of larger numbers of organisms on fingernails compared to freshly polished or natural nails. Also, dark colored nail polish may prevent dirt and debris under fingernails from being seen and removed (Baumgardner et al 1993).

Jewelry

Although several studies have shown that skin under rings is more heavily colonized than comparable areas of skin on fingers without rings (Jacobson et al 1985), at the present time it is not known whether wearing rings results in greater transmission of pathogens. It is suggested that surgical team members not wear rings because it may be more difficult for them to put on surgical gloves without tearing them.

REFERENCES

Alvarado CJ. 2000. *The Science of Hand Hygiene: A Self-Study Monograph*. University of Wisconsin Medical School and Sci-Health Communications. March.

Baumgardner CA et al. 1993. Effects of nail polish on microbial grown of fingernails. *AORN J* 58(4): 84–88.

Boyce JM and D Pittet. 2002. Guidelines for hand hygiene in healthcare settings: recommendations of the Healthcare Infection Control Practices Advisory Committee and the HICPAC/SHSA/APIC/IDSA Hand Hygiene Task Force. *Infect Control Hosp Epidemiol* 23(Suppl): S3–S40. Available at: http://www.cdc.gov/handhygiene.

Boyce JM. 1999. It is time for action: Improving hand hygiene in hospitals (editorial). *Ann Intern Med* 130(2): 153–155.

Centers for Disease Control (CDC). 1989. Guidelines for prevention of transmission of human immunodeficiency virus and hepatitis virus to health care and public-safety workers. *MMWR* 38(S-6): 5–6.

Deshmukh N, JW Kramer and SI Kjellberg.1996. A comparison of 5-minute povidone-iodine scrub and a 1-minute povidone-iodine scrub followed by alcohol foam. *Mil Med* 163(3): 145–147.

Dineen P. 1966. The use of a polyurethane sponge in surgical scrubbing. *Surg Gynecol Obstet* 123(3): 595–598.

Doebbeling BN et al. 1988. Removal of nosocomial pathogens from the contaminated glove. *Arch Intern Med* 109(5): 394–398.

Girou E et al. 2002. Efficacy of handrubbing with alcohol based solution versus standard handwashing with antiseptic soap: Randomized clinical trial. *BMJ* 325(7360): 362–365.

Hedderwick SA et al. 2000. Pathogenic organisms associated with artificial fingernails worn by healthcare workers. *Infect Control Hosp Epidemiol* 21(8): 505–509.

Jacobson G et al. 1985. Handwashing: Ring-wearing and number of microorganisms. *Nurs Res* 34(3): 186–188.

Kikuchi-Numagami K et al. 1999. Irritancy of scrubbing up for surgery with or without a brush. *Acta Derm Venereol* 79(3): 230–232.

Korniewicz D et al. 1990. Leakage of virus through used vinyl and latex examination gloves. *J Clin Microbiol* 28(4): 787–788.

Kotilainen HR et al. 1989. Latex and vinyl examination gloves. *Arch Intern Med* 149(12): 2749–2753.

Larson E et al. 2001. Comparison of different regimens for surgical hand preparation. *AORN J* 73(2): 412–432.

Larson E et al. 2000. An organizational climate intervention associated with increased handwashing and decreased nosocomial infections. *Behav Med* 26(1): 14–22.

Larson E. 1995. APIC guidelines for handwashing and hand antisepsis in health care settings. *Am J Infect Control* 23(4): 251–269.

Larson E et al. 1990. Alcohol for surgical scrubbing? *Infect Control Hosp Epidemiol* 11(3): 139–143.

Larson E. 1988. Guideline for use of topical antimicrobial agents. *Am J Infect Control* 16(6): 253–266.

McCormick RD et al. 2000. Double-blind, randomized trial of scheduled use of a novel barrier cream and an oil-containing lotion for protecting the hands of health care workers. *Am J Infect Control* 28(4): 302–310.

McGinley KJ, EL Larson and JJ Leydon. 1988. Composition and density of microflora in the subungual space of the hand. *J Clin Microbiol* 26(5): 950–953.

Moolenaar RL et al. 2000. A prolonged outbreak of *Pseudomonas aeruginosa* in a neonatal intensive care unit: did staff fingernails play a role in disease transmission? *Infect Control Hosp Epidemiol* 21(2): 80–85.

Muto CA et al. 2000. Hand hygiene rates unaffected by installation of dispensers of a rapidly acting hand antiseptic *Am J Infect Control* 28(3): 273–276.

Olsen RJ et al. 1993. Examination gloves as barriers to hand contamination in clinical practice. *JAMA* 270(3): 350–353.

Pereira LJ, GM Lee and KJ Wade. 1997. An evaluation of five protocols for surgical handwashing in relation to skin condition and microbial counts. *J Hosp Infect* 36(1): 49–65.

Pereira LJ, GM Lee and KJ Wade. 1990. The effect of surgical handwashing routines on the microbial counts of operating room nurses. *Am J Infect Control* 18(6): 354–364.

Pierce M. 1990. Personal reference. Johnson and Johnson Medical, Inc., Research Division: Arlington, TX.

Pittet D et al. 2000. Effectiveness of a hospital-wide programme to improve compliance with hand hygiene. *Lancet* 356(9238): 1307–1312.

Reingold Al, MA Kane and AW Hightower. 1988. Failure of gloves and other protective devices to prevent transmission of hepatitis B virus to oral surgeons. *JAMA* 259(17): 2558–2560.

Rutala WA. 1996. APIC guideline for selection and use of disinfectants. *Amer J Infect Control* 24(4): 313–342.

Semmelweiss IP. 1861. *Die Aetologie, der Begriff und die Prophylaxis Des Kindbettfiebers.* Pest: CA Hattleberi's Verlags-Expeditions.

Tenorio AR et al. 2001. Effectiveness of gloves in the prevention of hand carriage of vancomycin-resistant enterococcus species by health care workers after patient care. *Clin Infect Dis* 32(5): 826–829.

The Economist. 2002. How to save a million lives. 4 July (print edition).

Tibballs J. 1996. Teaching hospital medical staff to handwash. *Med J Aust* 164(7): 395–398.

FOUR

GLOVES

KEY CONCEPTS you will learn in this chapter include:

- When gloves should be worn
- Which type of glove to use
- What the glove requirements for clinical procedures are
- Glove use DOs and DON'Ts

BACKGROUND

Hand hygiene, coupled with the use of protective gloves, is a key component in minimizing the spread of disease and maintaining an infection-free environment (Garner and Favero 1986). In addition, understanding when sterile or high-level disinfected gloves are required and, equally important, *when they are not*, can reduce costs while maintaining safety for both patients and staff.

Until about 15 years ago, healthcare workers wore gloves for three reasons:

1. To reduce the risk of staff acquiring bacterial infections from patients.
2. To prevent staff from transmitting their skin flora to patients.
3. To reduce contamination of the hands of staff by microorganisms that can be transmitted from one patient to another (cross-contamination).

Furthermore, gloves were primarily worn only by staff caring for patients infected with certain pathogens or exposed to patients with high risk of hepatitis B.

Since 1987 and the emergence of the AIDS epidemic, a dramatic increase in glove use by all types of healthcare staff has occurred in an effort to prevent transmission of HIV and other bloodborne and body fluid viruses from patients to staff. As a result, disposable examination and surgical gloves are the item of personal protective equipment most frequently used by healthcare providers today. In the US, for example, glove usage has grown from 1.4 billion pairs in 1988 to 8.3 billion in 1993 (NIOSH 1997).

WHEN TO WEAR GLOVES

Although the effectiveness of gloves in preventing contamination of healthcare workers' hands has been repeatedly confirmed (Tenorio et al 2001), **wearing gloves does not replace the need for handwashing**. For example, even the best quality latex surgical gloves may have small, inapparent defects, gloves may be torn during use and hands can become contaminated during removal (Bagg, Jenkins and Barker 1990; Davis 2001).

Remember: Wash hands or use an antiseptic handrub before putting on gloves and after removing them.

Depending on the situation, clean **examination** or **utility** gloves should be worn by all staff when:

- there is reasonable chance of hand contact with blood or other body fluids, mucous membranes or nonintact skin;
- they perform invasive medical procedures (e.g., inserting vascular devices such as peripheral venous lines); or
- they handle contaminated waste items or touch contaminated surfaces.

Note: Examination gloves should be changed as soon as possible when visibly soiled, torn or punctured.

A separate pair of gloves must be used for each patient to avoid cross-contamination (CDC 1987). Wearing the same pair of gloves and washing gloved hands between patients or between dirty to clean body site care is not a safe practice. Doebbeling and colleagues (1988) recovered significant amounts of bacteria on the hands of staff who were just washing their gloved hands, not changing gloves between patients.

What to Do When Supplies of Gloves Are Limited

Hospital and clinic managers, and supervisors as well, should first check to be sure staff are not wearing gloves when they are not needed (i.e., for activities such as taking a patient's blood pressure, using the telephone or writing in a chart, and that do not involve contact with blood or other potentially infectious materials). In addition, when resources are limited and examination gloves are in short supply, soiled disposable surgical gloves can be reprocessed for reuse if they are:

- decontaminated by soaking in 0.5% chlorine solution for 10 minutes,
- washed and rinsed, and
- sterilized (by autoclaving) or high-level disinfected (by steaming).[1]

Do not reprocess gloves that are cracked, peeling or have detectable holes or tears (Bagg, Jenkins and Barker 1990).

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[1] In the past, boiling has been recommended as a method for HLD of surgical gloves; however, it is difficult to dry gloves without contaminating them using this method. Because steaming is easier to do and equally effective, it is the recommended method for HLD of surgical gloves (see **Appendix C**).

Where utility gloves are not available, putting on two pairs of examination or reprocessed surgical gloves (double gloving) provides some protection for cleaning staff and for staff handling and disposing of contaminated medical waste.

TYPES OF GLOVES

There are three types of gloves used in healthcare facilities: surgical, examination and utility or heavy-duty household gloves:

1. **Surgical gloves** should be used when performing invasive medical or surgical procedures.
2. **Examination gloves** provide protection to healthcare workers when performing many of their routine duties.
3. **Utility** or **heavy-duty household gloves** should be worn for processing instruments, equipment and other items; for handling and disposing of contaminated waste; and when cleaning contaminated surfaces.

The best **surgical gloves** are made of latex rubber, because of rubber's natural elasticity, sensitivity and durability and it provides a comfortable fit. Because of the increasing problem of latex allergy, a new synthetic rubber-like material called "nitrile," which has properties similar to latex, has been developed. Nitrile gloves are less likely to cause allergic reactions. In many countries, the only type of **examination gloves** usually available are made of vinyl, a synthetic material that is less expensive than latex rubber. Because vinyl is inelastic (does not stretch like latex), the gloves are loose-fitting and can tear easily. Better quality examination gloves are made from latex or nitrile and can be found in medical supply stores in most countries. Because **utility gloves** are made of thick rubber, which is much less flexible and sensitive,e they provide maximum protection as a barrier.

All types of examination gloves are very thin and should not be reprocessed for reuse (Korniewicz et al 1990).

The advantages and disadvantages of different types of gloves are described in **Table 4-1**.

Table 4-1. Advantages and Disadvantages of Different Types of Gloves

TYPE OF GLOVE	ADVANTAGES	DISADVANTAGES
Sterile or High-Level Disinfected Surgical Gloves[a]: Use for all procedures involving contact with tissue deep under the skin (e.g., cesarean section or laparotomy).	Gloves are sized to fit, permitting greater movement during surgical procedures.	Expensive; do not use for tasks where other types of gloves can be worn.
Examination Gloves: Use for contact with mucous membranes and nonintact skin (e.g., pelvic examination).	Inexpensive exam gloves are one quarter to one third the cost of surgical gloves and usually are available in most countries.	Usually, only small, medium and large sizes; may not be available in every country. When exam gloves are not available, used latex surgical gloves may be washed and steamed for reuse in patient care tasks requiring exam gloves.
Utility or Heavy-Duty Household Gloves: Use when handling used instruments and equipment that may have come in contact with blood or body fluids and for handling medical waste and linens.	Inexpensive; can be rewashed and reused many times. The thick rubber surface helps to protect cleaning personnel and waste handlers.	Not available in every country. If not available, double gloving using either new examination or reprocessed surgical gloves provides some protection.

[a] When surgical gloves are reused, they must be checked carefully for tears or cuts before final processing (Bagg, Jenkins and Barker 1990).

Adapted from: Tietjen, Cronin and McIntosh 1992.

Examination Gloves

Deciding which type of examination glove is best for a task (if a choice is available) should be determined by the degree of risk of exposure (low or high risk) to blood or potentially infected body fluids, the length of the procedure and possibility of allergy to latex or, rarely, nitrile.

- **Vinyl** examination gloves are the least expensive of the three types generally available. They are good for short tasks that involve minimal stress on the glove and low risk of exposure. They are loose-fitting (baggy), have limited elasticity and tear easily. Suggested use would be for briefly suctioning endotracheal secretions, emptying emesis basins and removing an IV line. (If they are the only type of examination glove available and the risk of exposure to blood and body fluids is high, change them frequently and consider double gloving.)
- **Natural rubber latex** examination gloves provide the best protection. They are preferred for surgical procedures and tasks of moderate to high risk such as exposure to blood or potentially contaminated body fluids. They should not be used by staff with known or suspected allergy to latex or for prolonged (>1 hour) contact with high-level disinfectants such as glutaraldehyde (may cause loss of effectiveness due to breakdown of latex).
- **Nitrile** examination gloves are the preferred choice for staff with latex allergy and may be used for activities of moderate to high risk. Nitrile gloves have many of the same characteristics as latex but have better resistance to oil-based products. Staff with known allergy to nitrile compounds should not use nitrile gloves.

Note: When using latex rubber gloves, do not use hand cream or lotions that contain mineral oil, petroleum jelly (Vaseline) or lanolin to protect your hands, because they may cause the gloves to break down within minutes.

GLOVE REQUIREMENTS FOR CLINICAL PROCEDURES

Listed in **Table 4-2** are common medical and surgical procedures that may require the use of protective gloves and the type of glove and or processing required. Sterile disposable surgical gloves always can be used, but because of their high cost should only be used when necessary. If the risk of endospores is not high (e.g., cesarean section or laparotomy), high-level disinfected surgical gloves are an acceptable alternative. (See **Chapter 1** for discussion.)[2]

Instructions are provided in **Appendix C** for how to process surgical gloves and either sterilize or high-level disinfect them, and how to store them safely.

Table 4-2. Glove Requirements for Common Medical and Surgical Procedures

TASK OR ACTIVITY	ARE GLOVES NEEDED?	PREFERRED GLOVES[a]	ACCEPTABLE GLOVES
Blood pressure check	No		
Temperature check	No		
Injection	No		
Blood drawing	Yes	Exam[b]	HLD Surgical[d]
IV insertion and removal	Yes	Exam[b]	HLD Surgical[d]
Pelvic examination	Yes	Exam	HLD Surgical[d]
IUD insertion (loaded in sterile package and inserted using no-touch technique)	Yes	Exam	HLD Surgical[d]
IUD removal (using no-touch technique)	Yes	Exam	HLD Surgical[d]
Manual vacuum aspiration (using no-touch technique)	Yes	Exam	HLD Surgical[d]
Norplant implants insertion and removal	Yes	Sterile Surgical[c]	HLD Surgical[d]
Vaginal delivery	Yes	Sterile Surgical[c]	HLD Surgical[d]
Cesarean section or laparotomy	Yes	Sterile Surgical[c]	HLD Surgical[d]
Vasectomy or laparoscopy	Yes	Sterile Surgical[c]	HLD Surgical[d]
Handling and cleaning instruments	Yes	Utility	Exam or HLD Surgical[d]
Handling contaminated waste	Yes	Utility	Exam or HLD Surgical[d]
Cleaning blood or body fluid spills	Yes	Utility	Exam or HLD Surgical[d]

[a] Although **sterile gloves** may be used for any surgical procedure, they are **not** always required. In some cases, examination or HLD surgical gloves are equally safe and less expensive.
[b] This includes new, "never" used individual or bulk-packaged examination gloves (as long as boxes are stored properly).
[c] When sterilization equipment (autoclave) is not available, high-level disinfection is the **only** acceptable alternative.
[d] Reprocessed surgical gloves.
Adapted from: Tietjen, Cronin and McIntosh 1992.

[2] Martin et al (1988) has reported that reprocessing surgical gloves more than three times usually is not cost-effective.

ACCIDENTAL CONTAMINATION OF STERILE OR HIGH-LEVEL DISINFECTED SURGICAL GLOVES

There are several ways to contaminate sterile or high-level disinfected surgical gloves:

Remember: Surgical staff wearing sterile or high-level disinfected gloves should be careful **not** to contaminate gloved hands inadvertently by touching nonsterile items and unprepped skin or mucous membranes.

- tearing or puncturing the glove,
- touching any nonsterile or high-level disinfected object with the glove, or
- touching the outside of a glove with an ungloved hand.

Regloving after contamination. To reglove after contaminating a glove during a surgical procedure:

- Remove contaminated glove by the cuff and, if reusing, place it in a 0.5% chlorine solution for decontamination; otherwise, put in waste container.

Sterile Glove

- Have the circulating nurse open the sterile glove pack, laying the glove package on a clean surface.
- Pick up the sterile glove with the gloved hand and put on the replacement glove in the usual manner.

Alternatively:

- Have the circulating nurse open the sterile glove package; then have the surgical assistant or scrub nurse, who is gloved, remove a sterile glove and hold the glove open by the cuff.[3] Put hand into the glove without touching the outside of the glove.
- Adjust the glove after the surgical assistant or scrub nurse lets go of the cuff (Sorensen and Luckman 1979).

High-Level Disinfected Glove

- Have the circulating nurse pick up the replacement glove with high-level disinfected forceps.
- Grasp the replacement glove by the turned-down cuff and put on the glove in the usual manner.

Alternatively:

- Have the circulating nurse remove a replacement glove from the high-level disinfected container with forceps. Have the surgical assistant,

[3] If the assistant or scrub person's gloves are contaminated with blood or body fluids, have someone with uncontaminated sterile gloves pick up and hold the replacement sterile glove.

who is gloved, take the glove and hold it open by the cuff.[4] Put hand into the glove without touching the outside of the glove.

- Adjust the glove after the surgical assistant or scrub nurse lets go of the cuff.

SOME DOs AND DON'Ts ABOUT GLOVES

- **Do** wear the correct size glove, particularly surgical gloves. A poorly fitting glove can limit your ability to perform the task and may be damaged (torn or cut) more easily.
- **Do** change surgical gloves periodically during long cases as the protective effect of latex rubber gloves decreases with time and inapparent tears may occur.
- **Do** keep fingernails trimmed moderately short (less than 3 mm or 1/8 inch beyond the finger tip) to reduce the risk of tears.
- **Do** pull gloves up over cuffs of gown (if worn) to protect the wrists.
- **Do** use water-soluble (nonfat-containing) hand lotions and moisturizers often to prevent hands from drying, cracking and chapping due to frequent handwashing and gloving.
- **Don't** use oil-based hand lotions or creams, because they will damage latex rubber surgical and examination gloves.
- **Don't** use hand lotions and moisturizers that are very fragrant (perfumed) as they irritate the skin under gloves.
- **Don't** store gloves in areas where there are extremes in temperature (e.g., in the sun, or near a heater, air conditioner, ultraviolet light, fluorescent light or X-ray machines). These conditions may damage the gloves (cause breakdown of the material they are made of), thus reducing their effectiveness as a barrier.

ALLERGIC REACTIONS TO GLOVES

Allergic reactions to latex rubber gloves are being increasingly reported among healthcare workers of all types, including housekeepers, laboratory workers and dentists. (Allergic reactions to nitriles also occur, but less frequently.) If possible, nonlatex (nitrile) or low-allergen latex gloves should be used if allergy is suspected. In addition, wearing powder-free gloves is recommended. (Powdered gloves may result in more reactions because the powder from the gloves carries the latex particles in the air.) If this is not possible, then wearing cloth or vinyl gloves beneath latex gloves may help to prevent skin sensitization. It will not, however, prevent sensitization of the mucous membranes of the eyes and nose if these gloves are powdered (Garner and HICPAC 1996).

[4] If the assistant or scrub person's gloves are contaminated with blood or body fluids, have someone with uncontaminated high-level disinfected gloves pick up and hold the replacement sterile glove.

For most sensitized people, the symptoms are skin rashes, runny nose and itchy eyes that may persist or get progressively worse (i.e., cause breathing problems such as asthma). An allergic reaction to latex can develop within 1 month of use. Even in people who are susceptible, however, reactions generally take longer to develop (within 3–5 years) and may not develop for as long as 15 years (Baumann 1992). No therapy or desensitization exists for latex allergy; therefore, the only option is to avoid contact.

REFERENCES

Bagg J, S Jenkins and GR Barker. 1990. A laboratory assessment of the antimicrobial effectiveness of glove washing and re-use in dental practice. *J Hosp Infect* 15(1): 73–82.

Baumann MA. 1992. Protective gloves. *Int Dent J* 42(3): 170–180.

Centers for Disease Control (CDC). 1987. Recommendations for prevention of HIV transmission in healthcare settings. *MMWR* 36(Suppl 2): 1S–18S.

Davis MS. 2001. Choices of effective personal protective equipment, in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta, pp 39–48.

Doebbeling BN et al. 1988. Removal of nosocomial pathogens from the contaminated glove. *Ann Intern Med* 109(5): 394–398.

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80 and *Am J Infect Control* 24(1): 24–52.

Garner JS and MS Favero. 1986. CDC guideline for handwashing and hospital environmental control, 1985. *Infect Control* 7(4): 231–243.

Korniewicz D et al. 1990. Leakage of virus through used vinyl and latex examination gloves. *J Clin Microbiol* 28(4): 787–788.

Martin MV et al. 1988. A physical and microbial evaluation of the re-use of non-sterile gloves. *Brit Dent J* 165(12): 421.

National Institute for Occupational Safety and Health, Department of Health and Human Services (NIOSH). 1997. *NIOSH Alert: Preventing Allergic Reaction to Nature Rubber Latex in the Workplace*. No. 97: 135.

Sorensen KC and J Luckman. 1979. *Basic Nursing: A Psychophysiologic Approach*. WB Saunders Co.: Philadelphia, pp 934–938.

Tenorio AR et al. 2001. Effectiveness of gloves in the prevention of hand carriage of vancomycin-resistant enterococcus species by health care workers after patient care. *Clin Infect Dis* 32(5): 826–829.

Tietjen LG, W Cronin and N McIntosh. 1992. Handwashing and gloving, in *Infection Prevention Guidelines for Family Planning Service Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 13–20.

FIVE

PERSONAL PROTECTIVE EQUIPMENT AND DRAPES

KEY CONCEPTS you will learn in this chapter include:

- Which personal protective equipment (PPE) and practices are effective
- What the limitations of PPE are
- What the various types of drapes are
- How to use drapes appropriately

BACKGROUND

Healthcare workers are confronted each day with the difficult task of working safely within a hazardous environment. Today, the most common occupational risk faced by healthcare personnel is contact with blood and body fluids during routine patient care. This exposure to pathogens increases their risk for serious infection and possible death. Health workers in some occupational settings, such as surgery and delivery rooms, have a higher risk of exposure to these pathogens than in all other departments combined (Gershon and Vlahov 1992; Gershon and Zirkin 1995). Because of this increasing risk, better infection prevention guidelines and practices are needed to protect staff working in these areas. Moreover, staff members who know how to protect themselves from blood and body fluid exposures and consistently use these measures will also help protect their patients.

While there is a growing awareness of the seriousness of AIDS, as well as hepatitis C, and how they are acquired in the workplace, many healthcare staff do not perceive themselves to be at risk. Moreover, even those that do perceive the risk do not regularly use protective equipment such as gloves, or other practices (e.g., handwashing) available to them. This is due in part to a mistaken belief that AIDS is largely confined to certain "at-risk" groups—sex workers, IV drug users or homosexuals. Although this may have been true several years ago, in 2002 WHO estimated that more than 40 million people were infected with HIV around the world and that the virus is increasingly affecting the heterosexual population and spreading to nonurban areas.

Ongoing research has identified several psychosocial and organizational factors that may contribute to lack of compliance by healthcare staff. The most important of these are perceived to be:

- poor safety conditions for staff working in hospitals and clinics, and
- conflict of interest between providing the best patient care and protecting oneself from exposure (Gershon 1996).

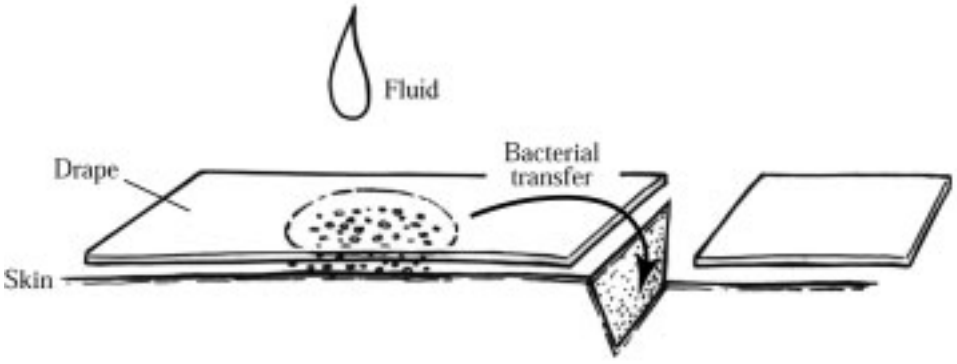
Finally, a study by the Institute of Medicine (1996) in the US implicated insufficient staffing and/or staff lacking the required knowledge and skills to meet the increased workload as important factors contributing to work-related injuries of nurses in hospitals. This situation also exists in most countries with limited resources as well.

PERSONAL PROTECTIVE EQUIPEMENT

Protective barriers, now commonly referred to as personal protective equipment (PPE), have been used for many years to protect patients from microorganisms present on staff working in the healthcare setting. More recently, with the emergence of AIDS and HCV and the resurgence of tuberculosis in many countries, use of PPE now has become important for protecting staff as well.

While some PPE, such as clean examination gloves, are extremely important in reducing the risk of transmission, others (e.g., cloth caps and shoe covers) continue to be used without convincing evidence of their effectiveness (Larson et al 1995). In fact, some common practices, such as having all staff in the operating room, not just the surgical team, wear masks, may increase costs while providing minimal, if any, protection to patients (Mitchell 1991). In addition, to be effective, PPE must be used correctly. For example, surgical gowns and drapes have been shown to prevent wound infection **only** when dry. When wet, cloth acts as a wick or sponge to draw bacteria from skin or equipment up through the fabric that can then contaminate a surgical wound (**Figure 5-1).**

Figure 5-1. Bacterial Transfer Through Fabric



As a consequence, hospital administrators, supervisors and healthcare workers need to be aware not only of the benefits and limitations of

specific PPE, but also of the actual role PPE play in preventing infection so that they can use them effectively and efficiently.

What Is Personal Protective Equipment?

Personal protective equipment includes: gloves, masks/respirators, eyewear (face shields, goggles or glasses), caps, gowns, aprons and other items. In many countries caps, masks, gowns and drapes are made of cloth or paper. The most effective barriers, however, are made of treated fabrics or synthetic materials that do not allow water or other liquids (blood or body fluids) to penetrate them. These fluid-resistant materials are not, however, widely available because they are expensive. Lightweight cotton cloth (with a thread count of 140/inch^2) is the material most commonly used for surgical clothing (masks, caps and gowns) and drapes in many countries. Unfortunately, lightweight cotton does not provide an effective barrier because moisture can pass through it easily, allowing contamination. Denims, canvas and heavy twill, on the other hand, are too dense for steam penetration (i.e., they cannot be sterilized), are hard to wash and take too long to dry. When fabric is used, it should be white or light in color in order to show dirt and contamination easily.

Caps, masks or drapes made from paper should never be reused because there is no way to properly clean them. **If you can't wash it, don't reuse it!**

Examples of how PPE can reduce the risk of spreading microorganisms and who (patients, staff or the community) the equipment protects are shown in **Table 5-1.**

In the following sections, the PPE that has proven to be effective is described as well as some commonly used items not shown to be effective.

Types of Personal Protective Equipment

Gloves protect hands from infectious materials and protect patients from microorganisms on staff members' hands. They are the most important physical barrier for preventing the spread of infection, but they must be changed between each patient contact to avoid cross-contamination. For example, examination gloves should be worn when handling blood, body fluids, secretions and excretions (except sweat), contaminated surfaces or equipment, and when touching nonintact skin or mucous membranes. (The appropriate use of gloves is discussed in detail in **Chapter 4**.)

Remember: Wearing gloves does **not** replace handwashing or use of antiseptic handrubs.

Masks should be large enough to cover the nose, lower face, jaw and facial hair (**Figure 5-2**). They are worn in an attempt to contain moisture droplets expelled as health workers or surgical staff speak, cough or sneeze, as well as to prevent accidental splashes of blood or other contaminated body fluids from entering the health workers' nose or

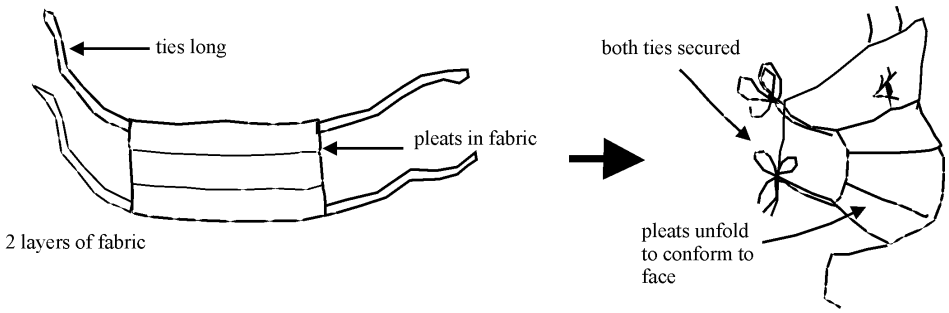
mouth. Unless the masks are made of fluid-resistant materials, however, they are not effective in preventing either very well.

Table 5-1. How Personal Protective Equipment Blocks the Spread of Microorganisms

WHERE MICROORGANISMS ARE FOUND	HOW MICROORGANISMS ARE SPREAD	BARRIERS TO STOP THE SPREAD OF MICROORGANISMS	WHO THE BARRIER PROTECTS
Healthcare staff			
hair and scalp	shedding skin or hair	cap	patient
nose and mouth	coughing, talking	mask	patient
body and skin	shedding skin or hair	scrubsuit, covergown	patient
hands	touching	gloves, handwashing or waterless antiseptic handrub	patient
Patient's mucous membranes and nonintact skin	touching	gloves	patient and staff
Patient's blood and body fluids	splashing or spraying	gloves, eyewear, mask, drapes, apron	staff
	touching (contact)	instrument processing	patient
		utility gloves,	staff
	accidental exposure with contaminated needles and scalpel blades	protective footwear, decontamination and disposal; use a Safe or Neutral Zone during surgery	staff
	infectious waste	utility gloves, plastic bags and disposal	staff and community
Patient's unprepped skin	touching	skin prep, drapes, gloves	patient
Clinic or hospital environment	touching	gloves, handwashing	staff and their family
		dressings	staff and community

Masks are made from a variety of materials ranging from lightweight cotton, gauze or even paper to synthetics, some of which are fluid-resistant. Masks made from cotton or paper are very comfortable but not fluid-resistant or effective as a filter. Masks made from synthetics can provide some protection from large-particle droplets (> 5 µm in size) spread by coughs or sneezes from a healthcare worker who is close (less than 3 feet/1 meter) to a patient. They are, however, somewhat uncomfortable to wear (difficult to breath through). Even the best surgical masks, however, are not designed to provide a tight enough fit (face seal) to prevent air leakage around the edges. Thus, they do not effectively filter inhaled air (Chen and Welleke 1992) and should no longer be recommended for that purpose.

Figure 5-2. Masks



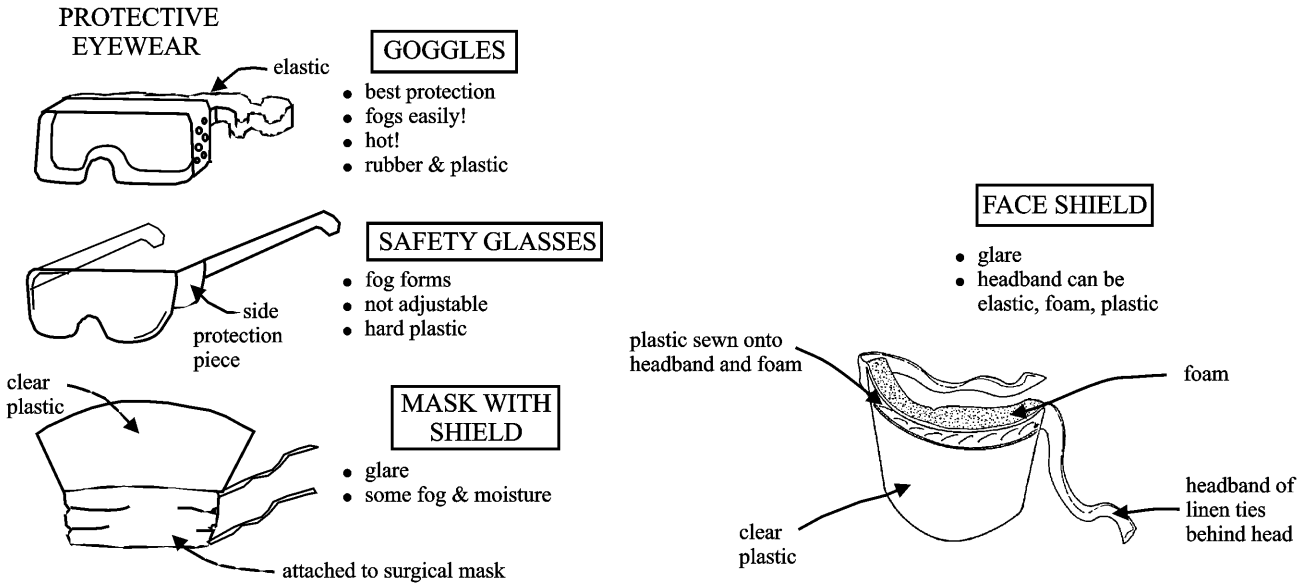
When removing, handle masks by the strings as the center of the mask contains the most contamination (Rothrock, McEwen and Smith 2003).

The true need for all operating room staff to wear a surgical mask as a means of preventing wound infection is questionable. Study results are conflicting, but even the authors of those showing no increase in wound infection rates acknowledge that masks should be worn by the surgeon and all staff who are scrubbed, in case of sneezing or coughing (Mitchell 1991). Thus, at present, the primary reason for wearing masks, especially those made of cotton gauze or paper (materials that are not fluid-resistant), is to provide some protection to the wearer from splashes or sprays of a patient's blood or potentially contaminated body fluids from entering the nose and mouth.

Respirators are specialized types of masks, called particulate respirators, that are recommended for situations in which filtering inhaled air is deemed important (e.g., for the care of a person with pulmonary tuberculosis). They contain multiple layers of filter material and fit the face tightly. They are considerably more difficult to breathe through and more expensive than surgical masks. Evidence that use of these specialized masks is effective is lacking.

Eyewear protects staff in the event of an accidental splash of blood or other body fluid by covering the eyes. Eyewear includes clear plastic goggles, safety glasses, face shields and visors. Prescription glasses or glasses with plain lenses also are acceptable (**Figure 5-3**). Masks and eyewear or face shields should be worn when performing any task where an accidental splash into the face is likely (e.g., performing cesarean section or vaginal delivery or when cleaning instruments). If face shields are not available, goggles or glasses and a mask can be used together.

Figure 5-3. Eyewear



Caps are used to keep the hair and scalp covered so that flakes of skin and hair are not shed into the wound during surgery. Caps should be large enough to cover **all** hair. While caps provide some protection to the patient, their primary purpose is to protect the wearer from blood or body fluid splashes and sprays.

Scrubsuits or covergowns are worn over, or instead of, street clothes. The main use of covergowns is to protect the healthcare workers' clothing. Scrubsuits usually consist of drawstring pants and a shirt. A V-neck shirt must not be cut so low as to slide off the wearer's shoulders or expose men's chest hair. There is little evidence that scrubsuits are needed during routine procedures when soiling of clothes is not likely (Goldman 1991). For example, in two studies, having personnel wear isolation gowns, caps and masks was not successful in reducing infection risk for patients as measured by infection or colonization (Donowitz 1986; Haque and Chagla 1989).

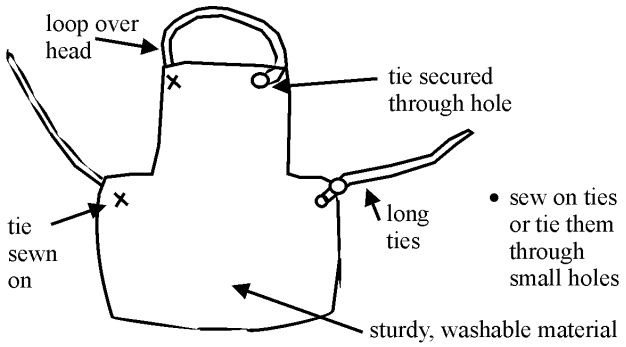
Surgical gowns were first used to protect patients from microorganisms present on the abdomen and arms of healthcare staff during surgery. Surgical gowns made of fluid-resistant materials do play a role in keeping blood and other fluids, such as amniotic fluid, off the skin of personnel, particularly in operating, delivery and emergency rooms. Lightweight cloth gowns, however, which are generally all that are available in most countries, offer little protection. Under these circumstances, if large spills occur, the best thing to do is shower or bathe as soon as possible after completing the operation or procedure. If surgical gowns are worn, sleeves should either taper gently toward the wrists or end with elastic or ties around the wrists. (Large, droopy sleeves invite accidental contamination.)

Remember: Do not lean on or rub up against draped areas, because bacteria penetrate even dry material easily due to the physical pressure exerted by leaning against the drapes.

In addition, the cuffs of the surgical gloves should completely cover the end of the sleeves.

Aprons made of rubber or plastic provide a waterproof barrier along the front of the health worker's body (**Figure 5-4**). An apron should be worn when cleaning or during a procedure in which blood or body fluid spills are anticipated (e.g., cesarean section or vaginal delivery). Aprons keep contaminated fluids off the healthcare worker's clothing and skin. In surgery, wearing a clean plastic apron over the scrubsuit will not only help prevent the surgeon or assistant from being exposed to blood or body fluids (e.g. amniotic fluid), but also prevent the surgeon's or assistant's abdominal skin from being a source of contamination to the patient (Moylan and Kennedy 1980).

Figure 5-4. Aprons



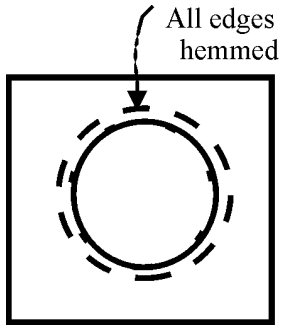
Footwear is worn to protect feet from injury by sharps or heavy items that may accidentally fall on them. For this reason, sandals, "thongs" or shoes made of soft materials (cloth) should not be worn. Rubber boots or leather shoes provide more protection, but they must be kept clean and free of contamination from blood or other body fluid spills. Shoe covers are unnecessary if clean, sturdy shoes are available for use **only** in the surgical area. One study suggests that cloth or paper shoe covers may increase contamination because they allow blood to soak through to shoes and they are often worn outside the operating room where they are then removed with ungloved hands (Summers et al 1992).

THE ROLE OF DRAPES

In many countries, drapes are usually made of hemmed linen squares of varying sizes. They are used to create an operative field around an incision, wrap instruments and other items for sterilization, cover tables in the operating room and keep clients warm during surgical procedures (OR Manager 1990a). The main types of drapes are:

- **Towel drapes** are used for drying hands, squaring off the operative site (several towel drapes are needed for this) and wrapping small instruments and syringes. They are often made of heavier cotton cloth than other linen items, which makes them somewhat more water-resistant.
- **Drapes** or **lap sheets** are used for covering the patient. They are large, usually made of lightweight cotton and provide only limited protection to patients or staff.
- **Site drapes** are made of cotton and have a circular opening in the center that is placed over the prepped operative site (**Figure 5-5**). These drapes are primarily intended for use with minor surgical procedures (small incisions).

Figure 5-5. Site Drape Sheet



- **Pack wrapper drapes**, large drapes that become a table cover when the sterile instrument pack is opened. This drape only needs to be large enough for wrapping the instruments and, when opened, to cover the tabletop completely.

Using Drapes for Surgical Procedures

Remember: Once a sterile drape touches the patient's skin, it is no longer sterile.

Using sterile **towel drapes** to create a work area around the incision limits the amount of skin that needs to be cleaned and prepped with antiseptic solution prior to placing the drapes. Although this area is often called the "sterile field," it is only briefly sterile. As shown in **Figure 5-1**, cloth drapes allow moisture to soak through them and can help spread organisms from skin, even after surgical cleansing with an antiseptic agent, into the incision. Thus, neither gloved hands (sterile or high-level disinfected) nor sterile or high-level disinfected instruments and other items should touch the towel drapes once they are in place. Because cloth drapes do not serve as an effective barrier, clean, dry towel drapes can be used if sterile towel drapes are not available.

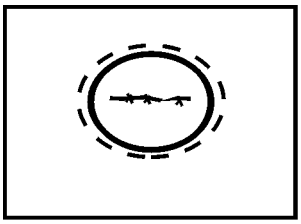
The way in which the operative site is prepared and draped depends on the type of procedure to be performed. The following guidelines for draping are designed to reduce overuse of costly sterile items and to avoid unnecessary draping:

- All drapes should be applied around a completely dry, widely prepped area.
- If sterile drapes are used, sterile or high-level disinfected surgical gloves should be worn when placing the drapes. (When putting drapes in place, care must be taken not to touch the patient's body with gloved hands.)
- Drapes should be handled as little as possible and should never be shaken or flapped. Always hold drapes above the area to be draped, and discard the drape if it falls below this area.

Minor Surgical Procedures (Norplant implants insertion or removal or minilaparotomy)

- Use a **site drape** that allows at least 5 cm (or 2 inches) of open skin around the incision (**Figure 5-6**). Alternatively, towel drapes can be used. (If sterile site or towel drapes are not available, clean, dry drapes can be used.)

Figure 5-6. Placing a Site Drape



site drape in place

- Place the hole in the drape over the prepped incision site and do not move it once it has touched the skin.
- If the site drape is not sterile, put on sterile or high-level disinfected gloves **after** placing the drape on the patient to avoid contaminating the gloves.

Major Surgical Procedures (laparotomy or cesarean section)

Remember: Lap sheets do not need to cover the entire patient.

- Use large **drapes** or **lap sheets** to cover the patient's body if it is necessary to keep her warm. These drapes do not need to be sterile because they will not be near the incision site (Belkin 1992). They should be clean and dry.

Remember: Sterile cloth drapes do not replace good **aseptic technique**.

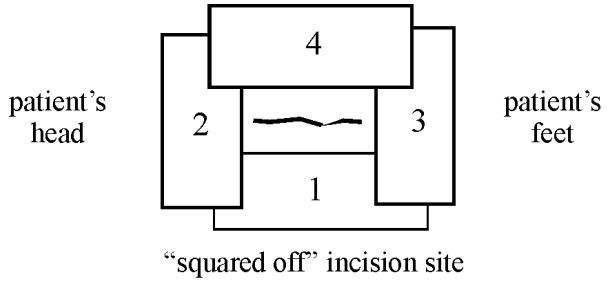
- After cleansing the skin with an antiseptic agent, place the towel drapes to square off the incision site (allow at least 5 cm, or 2 inches, of open skin around all sides of the proposed incision site).
- Begin by placing the towel drape closest to you to decrease the chance of contamination (**Figure 5-7**). Holding one side of the drape, allow the other side to touch the abdominal skin about 2 inches away from

the proposed incision site. Gently drop the rest of the drape onto the abdomen. Once in place, the drape should never be moved closer to the incision. It can, however, be pulled away from it.

- Place three additional drapes (2, 3 and 4) to square off the work area as shown in **Figure 5-7**.

Note: Avoid reaching across the incision site unless it has been draped.

Figure 5-7. Squaring Off a Work Area



- Use **nonperforating** towel clips to secure the corners of the towel drapes.

During Procedures

Do not use the patient's body or the draped area for placing instruments. Placing sterile or high-level disinfected instruments or other items on drapes, even if they were sterile initially, will contaminate them. Also, doing this may make the items harder to find and may cause them to fall off the operating room table if the patient moves. If an instrument stand (Mayo) covered with a sterile towel or drape is not available, a sterile or high-level disinfected plastic or metal instrument tray can be placed on the drape covering the patient and used to hold instruments during the procedure.

If a drape is torn or cut during a procedure, it should be covered with a new drape. Do not, however, place new drapes on top of a drape that has become wet. There is no evidence that this is effective in creating a barrier (OR Manager 1990b).

As drapes wear out and new drapes are needed, try to buy replacement drapes that have a high thread count.

See **Chapter 13** for information on processing linens (caps, gowns, masks and drapes).

MAKING THE WORKPLACE SAFER

Despite the limited success of educational programs aimed at changing healthcare worker behavior regarding use of PPE, primary prevention must continue to be the focus of future actions. To be more successful, efforts designed to make the workplace environment safer should be directed to all cadres of health workers—not just physicians and nurses. For example, in some countries, with the exception of operating room personnel, housekeeping staff have the highest rate of needlesticks injuries caused by used needles being incorrectly discarded in wastebaskets.

Improving compliance following educational and behavior change efforts can be enhanced if:

- There is consistent support by hospital administrators of the recommended safety efforts (e.g., identified deficiencies are corrected, dangerous practices are eliminated and staff are actively encouraged to seek inexpensive, doable solutions).
- Supervisors regularly provide feedback and reward appropriate behavior (e.g., handwashing between patient contacts).
- Role models, especially physicians and other senior staff and faculty, actively support recommended infection prevention practices and model appropriate behavior (Lipscomb and Rosenstock 1997).

Moreover, making the recommendations appropriate and easy for staff to use and monitor can lead to better compliance and health worker safety. Finally, because healthcare is a vitally important and rewarding profession, it is the responsibility of all healthcare professionals to help create a safer environment for patients and fellow workers.

REFERENCES

Belkin N. 1992. Barrier methods: Their influence on surgical wound infection. *AORN J* 55(6): 1521–1528.

Chen CC and K Welleke. 1992. Aerosol penetration through surgical masks. *Am J Infect Control* 20(4): 177–184.

Donowitz LG (ed). 1986. Failure of the cover gown to prevent nosocomial infection in pediatric intensive care unit. *Pediatrics* 77(1): 35–38.

Gershon R. 1996. Facilitator report: Bloodborne pathogens exposure among health care workers. *Am J Ind Med* 29(4): 418–420.

Gershon R and B Zirkin. 1995. Behavioral factors in safety training, in *Laboratory Safety, Principles and Practices*, 2nd ed. Flemming DO et al (eds). AMS Press: Washington, DC, pp 269–277.

Gershon R and D Vlahov. 1992. Assessing and reducing HIV risk to the critical care nurse. *Critical Care Nursing Currents* 3: (No 3).

Goldman DA. 1991. The role of barrier precautions in infection control. *J Hosp Infect* 18(Suppl A): 515–523.

Haque KN and AH Chagla. 1989. Do gowns prevent infections in neonatal intensive care units? *J Hosp Infect* 14(2): 159–162.

Institute of Medicine. 1996. *Nursing Staff in Hospitals and Nursing Home—Is It Adequate?* National Academy Press: Washington, DC.

Larson EL et al. 1995. APIC Guidelines for Infection Control Practice. Guidelines for handwashing and hand antisepsis in health care settings. *Am J Infect Control* 23(4): 251–269.

Lipscomb J and L Rosenstock. 1997. Health care workers: protecting those who protect our health. *Infect Control Hosp Epidemiol* 18(6): 397–399.

Mitchell NJ. 1991. Surgical facemasks in modern operating rooms—a costly and unnecessary ritual? *J Hosp Infect* 18(3): 239–242.

Moylan JS and BV Kennedy. 1980. The importance of gown and drape barriers in the prevention of wound infection. *Surg Gynecol Obstet* 151(4): 465.

OR Manager. 1990a. Draping controversy unveiled. *Today's OR Nurse* 12(8): 33.

OR Manager. 1990b. Draping: how much and which one? *Today's OR Nurse*12(9): 29.

Rothrock J, D McEwen and D Smith (eds). 2003. *Alexander's Care of the Patient in Surgery*, 12th ed. Mosby-Year Book, Inc.: St Louis, MO.

Summers PR et al. 1992. Blood-saturated operating-room shoe covers. *West J Med* 157(2): 184–185.

SIX

SURGICAL ANTISEPSIS

KEY CONCEPTS you will learn in this chapter include:

- What the causes of wound infections are
- What the safest and most effective antiseptics are
- How to use antiseptics and perform surgical antisepsis
- How to prevent contamination of antiseptics

BACKGROUND

Although considerable progress has been made in understanding the cause and prevention of surgical site infections during the past 100 years, postoperative wound infections (incisional and deep) remain a leading cause of nosocomial (hospital-acquired) infections, especially in developing countries. The vast majority of postoperative incisional or superficial wound infections are caused by microorganisms (usually bacteria or sometimes fungi) normally found on the patient's skin or from mucous membranes adjacent to the surgical site, and less often from other sites (e.g., nose, mouth or respiratory tract in abdominal operations). By contrast, microorganisms from the hands of the surgeon or assistant are seldom the cause of incisional surgical site infections (Galle, Homesley and Rhyne 1978), nor are organisms present in the operating room or on other surgical staff.

Preoperative surgical antisepsis consists of three processes (hand hygiene and gloving of surgical team members combined with applying an antiseptic agent to the surgical site) designed to block transmission of infectious agents into the surgical wound. The effectiveness of handwashing followed by briefly applying a waterless, alcohol-based antiseptic handrub or antiseptic solution in reducing the number of bacteria and fungi on hands has been amply documented (Galle, Homesley and Rhyne 1978; Larson et al 2001). In fact, one large, 10-year prospective study found no postoperative wound infections after 141 operations during which the surgeon's glove was punctured (Cruse and Foord 1980). In addition, preoperative skin preparation using an antiseptic agent, when done correctly, has been shown to effectively reduce both transient and resident skin flora, as well as infection rates (Platt and Bucknall 1984).

Whether a postoperative infection occurs depends on several risk factors, the most important being:

- number of microorganisms entering the wound;
- type and virulence (ability to cause disease) of the bacteria;
- strength of the patient's defense mechanisms (e.g., status of the immune system); and
- external factors, such as the patient being in the hospital several days before the surgery or duration of the surgery (>4 hours).[1]

Thus surgical antisepsis, by limiting the type and number of microorganisms transferred into the wound during surgery, plays an important, but not necessarily major, role in preventing postoperative wound infections.

DEFINITIONS

- **Antiseptic** or **antimicrobial agent (terms used interchangeably)**. Chemicals that are applied to the skin or other living tissue to inhibit or kill microorganisms (both transient and resident) thereby reducing the total bacterial count. Examples include alcohols (ethyl and isopropyl), dilute iodine solutions, iodophors, chlorhexidine and triclosan. (See **Appendix B** for complete listing of uses, effectiveness, advantages and disadvantages.)
- **Antisepsis**. Process of reducing the number of microorganisms on skin, mucous membranes or other body tissue by applying an antimicrobial (antiseptic) agent.

SELECTION OF ANTISEPTICS

While plain soap and clean water physically remove dirt and other material as well as some **transient** microorganisms from the skin, antiseptic solutions kill or inhibit almost all transient and many **resident** microorganisms, including most vegetative bacteria and many viruses. Antiseptics are designed to remove as many microorganisms as possible without damaging or irritating the skin or mucous membrane on which they are used. In addition, some antiseptic solutions have a **residual effect**, meaning their killing action continues for a period of time after they have been applied to skin or mucous membranes.

Many chemicals qualify as safe antiseptics. **Table 6-1** lists several recommended antiseptic solutions, their microbiologic activity and their potential uses. (The grading system used in this table is excellent, good, fair, and none.) The most frequently used antiseptics are chlorhexidine gluconate, which is contained in Hibitane®, Hibiscrub®, and iodophors such as Betadine® and Wescodyne®. Not listed in **Table 6-1** is Savlon®, which contains chlorhexidine and is available throughout the world, because it is largely sold as concentrated solution that is then diluted with water. In many countries, the concentration used is less than 1%, which is too low to be effective.

[1] The factors responsible for postoperative wound infections are discussed in more detail in **Chapter 23**.

Table 6-1. Antiseptics: Microbiologic Activities and Potential Uses

GROUP	ACTIVITY AGAINST BACTERIA							POTENTIAL USES			
	Gram-Positive	Most Gram-Negative	TB	Viruses	Fungi	Endospores	Relative Speed of Action	Affected by Organic Matter	Surgical Scrub	Skin Preparation	Comments
Alcohols (60–90% ethyl or isopropyl)	Excellent	Excellent	Excellent	Excellent	Excellent	None	Fast	Moderate	Yes	Yes	Not for use on mucous membranes Not good for physical cleaning of skin, no persistent activity
Chlorhexidine (2–4%) (Hibitane, Hibiscrub)	Excellent	Good	Fair	Excellent	Fair	None	Intermediate	Slight	Yes	Yes	Has good persistent effect Toxicity to ears and eyes
Iodine preparations (3%)	Excellent	Excellent	Excellent	Excellent	Good	Fair	Intermediate	Marked	No	Yes	Not for use on mucous membranes Can burn skin so remove after several minutes
Iodophors (7.5–10%) (Betadine)	Excellent	Excellent	Fair	Good	Good	None	Intermediate	Moderate	Yes	Yes	Can be used on mucous membranes
Para-chloro-metaxylenol (PCMX) (0.5–4%)	Good	Excellent	Fair	Good	Fair	Unknown	Slow	Minimal	No	Yes	Penetrates the skin and should not be used on newborns
Triclosan (0.2–2%)	Excellent	Good	Fair	Excellent	None	Unknown	Intermediate	Minimal	Yes	No	Acceptability on hands varies

Adapted from: Boyce and Pittet 2002; Olmsted 1996.

Although antiseptics are sometimes used as disinfectants (e.g., Savlon or Dettol®) for processing instruments and other inanimate objects, they are not designed for this use. They do not have the same killing power as chemical disinfectants (e.g., glutaraldehydes, hypochlorite and peroxides) and should not be used for this purpose (Rutala 1996).

Additional information, including advantages and disadvantages of commonly used antiseptics, is presented in **Appendix B**.

USE OF ANTISEPTICS

Hand Hygiene

Antimicrobial soaps or detergents are no more effective than plain soap and clean water in reducing the risk of infection when used for routine handwashing, provided the water quality is satisfactory (Pereira, Lee and Wade 1997). Water that contains large amounts of particulate matter (makes the water cloudy) or is contaminated (high bacteria count) should not be used for performing a surgical handscrub[2]. In addition, antimicrobial soaps are costly and are more irritating to the skin than plain soap. Detailed instructions for performing a surgical handscrub using either an antiseptic solution or antiseptic handrub are presented in **Chapter 3** and **Appendix A**.

Skin Preparation Prior to Surgical Procedures

Although skin cannot be sterilized, applying an antiseptic solution minimizes the number of microorganisms around the surgical wound that may contaminate and cause infection.

Instructions

STEP 1: **Do not shave hair** around the operative site. Shaving increases the risk of infection 5–10 fold because the tiny nicks in the skin provide an ideal setting for microorganisms to grow and multiply (Nichols 1991; Seropian and Reynolds 1971). If hair must be cut, **trim** the hair close to the skin surface with scissors immediately before surgery.

STEP 2: Ask the patient about **allergic reactions** (e.g., to iodine preparations) before selecting an antiseptic solution.

STEP 3: If the skin or external genital area is visibly soiled, gently wash it with soap and clean water and dry the area before applying the antiseptic.

Select the antiseptic solution from the following recommended products:

- Alcohol-based solutions (tinctures) of iodine or chlorhexidine
- Alcohols (60–90% ethyl, isopropyl or "methylated spirit") (see **Appendix B**)

[2] If tap water is cloudy, most particulates (debris and organic material) can be removed by filtering through four layers of moderately woven cotton cloth, such as cheese cloth or old sari material, before boiling or treating with dilute chlorine (sodium hypochlorite) solution (Colwell et al 2003; Huq et al 1996).

- Chlorhexidine gluconate (2–4%) (e.g., Hibitane, Hibiscrub, Hibiclens®)
- Chlorhexidine gluconate and cetrimide, various concentrations at least 2% (e.g., Savlon)
- Iodine (3%); aqueous iodine and alcohol-containing (tincture of iodine) products
- Iodophors (7.5–10%), various other concentrations (e.g., Betadine)
- Chloroxylenol (Para-chloro-metaxylenol or PCMX) (0.5–3.75%), various other concentrations (e.g., Dettol)

Note: **Do not allow the antiseptic to pool** underneath the client's body; this can irritate the skin.

STEP 4: Using dry, high-level disinfected forceps and new cotton or gauze squares soaked in antiseptic, thoroughly cleanse the skin.[3] Work from the operative site outward for several centimeters. (A circular motion from the center out helps to prevent recontamination of the operative site with local skin bacteria.)

STEP 5: Allow the antiseptic enough time to be effective before beginning the procedure. For example, when an iodophor is used, allow 2 minutes or wait until the skin is visibly dry before proceeding, because free iodine, the active agent, is only released slowly (see **Appendix B**).

INSTRUCTIONS FOR CERVICAL OR VAGINAL PREPARATION

For **cervical** and **vaginal antisepsis**, prior to inserting a uterine elevator for minilaparotomy or doing an endometrial biopsy, select an aqueous (water-based) antiseptic such as an iodophor (povidone-iodine) or 2–4% chlorhexidine gluconate (e.g., Hibiclens or Savlon if properly prepared). **Do not use alcohols or alcohol-containing preparations**, such as Dettol. Alcohols burn, and they also dry and irritate mucous membranes that in turn promote the growth of microorganisms. In addition, hexachlorophene (pHisoHex®) is neurotoxic (Larson 1988) and should not be used on mucous membranes, such as the vaginal mucosa, because it is readily absorbed (Larson 1995).

STEP 1: Ask the patient about **allergic reactions** (e.g., to iodine preparations) before selecting an antiseptic solution.

STEP 2: If the external genital area is visibly soiled, gently wash it with soap and clean water and dry the area before applying the antiseptic.

STEP 3: After inserting the speculum, apply antiseptic solution liberally to the cervix and vagina (two times). It is not necessary to prep the external genital area with antiseptic solution if it appears clean.

STEP 4: If an iodophor is used, allow time (2 minutes) before proceeding.

[3] The cotton or gauze swabs or pads do not need to be made up from sterile items. Clean, new (not reprocessed) cotton or gauze swabs can be used, because they do not contain harmful organisms and will be touching only noncritical (intact skin) and semicritical (mucous) membranes (Spaulding 1968).

Skin Preparation for Injections

According to WHO and its Safe Injection Global Network (SIGN), "swabbing of clean skin—with an antiseptic solution—prior to giving an injection is unnecessary," because in controlled trials no infections were noted. In addition, a review of microbiologic studies did not suggest that wiping the skin with an antiseptic before giving an intradermal, subcutaneous or intramuscular injection reduced the risk of infection (Hutin et al 2001).

> If the injection site is visibly soiled, wash the site with soap and water and dry with a clean towel, and then give the injection.[4]

STORING AND DISPENSING OF ANTISEPTICS

Contamination of **every** antiseptic agent has been documented. Microorganisms contaminating antiseptic solutions include *Staphylococcus epidermidis* and *aureus*, gram-negative bacilli, *Pseudomonas aeruginosa,* and some endospores. Contaminated antiseptics can cause subsequent infection when used for handwashing or preparing a client's skin. The following can prevent contamination of antiseptic solutions:

- Unless supplied commercially in small quantities, pour the antiseptic into a small, reusable container for daily use. This prevents evaporation and contamination. Make sure the correct name of the solution is on the container each time you refill it. **Do not store gauze or cotton wool in antiseptics because this promotes contamination.**
- Establish a routine schedule for preparing new solutions and cleaning reusable containers. (Solutions are at increased risk of becoming contaminated after 1 week of storage.) **Do not "top off" antiseptic dispensers.**
- **Wash** reusable containers thoroughly with soap and clean water, rinse with boiled water if available and **drip dry** before refilling.
- Label reusable containers with the date each time they are washed, dried and refilled.
- Concentrated antiseptic solutions should be stored in a cool, dark area. Never store them in direct sunlight or in excessive heat (e.g., upper shelves in a tin-roofed building).

[4] Patients receiving injections regularly (e.g., using DMPA for contraception) should be taught to wash the injection site (arm or buttocks) with soap and clean water just prior to coming to the clinic or receiving the injection at their home.

REFERENCES

Boyce JM and D Pittet. 2002. Guidelines for hand hygiene in healthcare settings: Healthcare infection control practices advisory committee and the HICPAC/SHSA/APIC/IDSA hand hygiene task force. *Infect Control Hosp Epidemiol* 23(12 Suppl): S3–S40. (Also can be accessed at www.cdc.gov/ncidod/nip/default.htm.)

Colwell RR et al. 2003. Reduction of cholera in Bangladeshi villages by simple filtration. *Proc Nat Acad Sci USA* 100(3): 1051–1055.

Cruse PJE and R Foord. 1980. The epidemiology of wound infection: A 10-year prospective study of 62,939 wounds. *Surg Clin North Am* 60(1): 27–40.

Galle PC, HD Homesley and AL Rhyne. 1978. Reassessment of the surgical scrub. *Surg Gynecol Obstet* 147(2): 215–218.

Huq A et al. 1996. A simple filtration method to remove plankton-associated *Vibrio cholera* in raw water supplies in developing countries. *Appl Environ Microbiol* 62(7): 2508–2512.

Hutin Y et al. 2001. Best infection control practices for intradermal, subcutaneous and intramuscular needle injections. World Health Organization (WHO), Safe Injection Global Network: Geneva.

Larson EL et al. 2001. Comparison of different regimens for surgical hand preparation. *AORN J* 73(2): 412–432.

Larson EL. 1995. APIC guidelines for handwashing and hand antisepsis in health care settings. *Am J Infect Control* 23(4): 251–269.

Larson EL. 1988. Guideline for use of topical antimicrobial agents. *Am J Infect Control* 16(6): 253–266.

Nichols RL. 1991. Surgical wound infection. *Am J Med* 91(Suppl 3B): 54S–64S.

Olmsted RN (ed). 1996. *Infection Control and Applied Epidemiology: Principles and Practices*. Association for Practitioners in Infection Control (APIC), Table 19-2. CV Mosby: St. Louis, MO.

Pereira LJ, GM Lee and KJ Wade. 1997. An evaluation of five protocols for surgical handwashing in relation to skin condition and microbial counts. *J Hosp Infect* 36(1): 49–65.

Platt J and RA Bucknall. 1984. An experimental evaluation of antiseptic wound irrigation. *J Hosp Infect* 5(2): 181–188.

Rutala WA. 1996. APIC guidelines for selection and use of disinfectants. *Am J Infect Control*, 24(4): 313–342.

Seropian R and BM Reynolds. 1971. Wound infections after preoperative depilatory versus razor preparation. *Am J Surg* 121(3): 251–254.

Spaulding EH (ed). 1968. Chemical disinfection of medical and surgical materials, in *Disinfection, Sterilization and Preservation.* Lawrence CA et al (eds). Lea & Febiger: Philadelphia, pp 437–446.

SEVEN

SAFE PRACTICES IN THE OPERATING ROOM

KEY CONCEPTS you will learn in this chapter include:

- Why the operating room is so risky for patients and staff
- Which instruments cause most injuries in the operating room and why
- How to avoid injuries from sharps
- How to manage exposure to blood and potentially contaminated body fluids

BACKGROUND

> **"The operating room is clearly one of the most hazardous environments in the healthcare delivery system. By definition, surgery is invasive. Instruments that are designed to penetrate patients' tissue can just as easily injure the provider. Blood is everywhere. Speed is essential. Emergencies can occur at any time and interrupt routines. Preventing injuries and exposures [to infectious agents] under these circumstances is indeed challenging!"**
>
> **—Julie Louise Gerberding, MD, MPH**
> ***Advanced Precautions for Today's OR*** **(Davis 2001a)**

In the past decade, awareness of the risk of exposure to blood and body fluids containing HIV, HBV and most recently HCV have created a new era in surgical infection prevention practices. Just as patients must be protected from wound contamination and infections, so must providers be protected from intraoperative injuries and exposure to patients' blood and other body fluids.

Preventing infections following an operation is a complex process that begins in the operating room by preparing and maintaining a safe environment for performing the surgery. Surgical aseptic techniques are designed to create such an environment by controlling the four main sources of infectious organisms: the patient, surgical staff, equipment and the operating room environment. Although the patient is often the source of surgical infections, the other three sources are important and should not be overlooked (see **Chapter 6**).

The science of safety in the surgical unit, whether it is located in a large specialty hospital or freestanding primary healthcare clinic, has not kept pace with the urgent need for prevention strategies. Although some of the

specific recommendations presented in this chapter have not been evaluated in clinical trials, they have been found over time to be worthwhile and merit further consideration. Fortunately, the effectiveness of many of these recommendations—HBV immunization, use of appropriate personal protective equipment when indicated (see **Chapter 5**), double gloving, sharps management and the use of blunt needles for suturing—is well-supported by data.

Specific techniques required to establish and maintain surgical asepsis and make the surgical environment safer include the following:

- **Patient considerations**: skin cleaning pre-operatively, skin antisepsis and wound covering (**Chapters 6** and **23**)
- **Surgical staff considerations**: hand hygiene (handwashing and/or handrub and handrubbing with waterless, alcohol-based antiseptic agents); use and removal of gloves and gowns (**Chapters 3 and 5**)
- **Equipment and room preparation considerations**: traffic flow and activity patterns as well as housekeeping practices (**Chapters 15** and **16**) and decontamination, cleaning and either sterilization or high-level disinfection of instruments, gloves and other items (**Chapters 10–12**)
- **Environmental considerations**: maintaining an aseptic operating field and using safer operating practices and techniques (**Chapter 7 and 15**)

Because traffic flow, equipment processing and room preparation requirements are discussed in other chapters, the focus of this chapter will be on improving the surgical environment (operating room), especially the practices and techniques that make surgery safer for both the patient and staff.

DEFINITIONS

- **Antisepsis**. Process of reducing the number of microorganisms on skin, mucous membranes or other body tissue by applying an antimicrobial (antiseptic) agent.
- **Asepsis** and **aseptic technique**: Combination of efforts made to prevent entry of microorganisms into any area of the body where they are likely to cause infection. The goal of asepsis is to **reduce to a safe level** or **eliminate** the number of microorganisms on both animate (living) surfaces (skin and tissue) and inanimate objects (surgical instruments and other items).
- **Surgical asepsis**. Preparation and maintenance of a reduced (safe) level of microorganisms during an operation by controlling four main sources of infectious organisms: the patient, personnel, equipment and the environment.

THE SURGICAL ENVIRONMENT

The operating room has special characteristics that increase the chance of accidents. For example, staff often use and pass sharp instruments without looking or letting the other person know what they are doing. The workspace is confined and the ability to see what is going on in the operative field for some members of the team (scrub nurse or assistant) may be poor. There is, moreover, the need for speed and the added stress of anxiety, fatigue, frustration and occasionally even anger. Finally, there is the fact that exposure to blood often occurs without the person's knowledge, usually not until the gloves are removed at the end of the procedure, which prolongs the duration of exposure. The fact that fingers are frequently the site of minor scratches and cuts further increases the risk of infection with bloodborne pathogens.

Which Instruments Cause Injuries

The vast majority of sharps injuries in hospitals occur in the operating room, and most are due to scalpel and suture-needle injuries, which is not surprising given that these are the two most frequently used sharps during operations. Many other items can also cause sharps injuries and glove tears resulting in exposure to blood. Some of the most important are:

- Hypodermic needles
- Wire sutures
- Laparoscopy and surgical drain trocars
- Orthopedic drill bits, screws, pins, wires and saws
- Needle point cautery tips
- Skin hooks and towel clips
- Sharp-pointed scissors and sharp-tipped mosquito forceps
- Dissecting forceps
- Sharp-toothed tenaculi

When Do Injuries Occur

Scalpel injuries most often occur when:

- Putting on and taking off the disposable blade
- Passing the scalpel hand to hand between team members
- Cutting (e.g., in using fingers to hold or spread tissue or cutting toward the fingers of the surgeon or assistant)
- Before and after using the scalpel: leaving it on the operative field, dropping it on your own or the assistant's foot, and reaching for scalpels sliding off the drapes
- Placing the scalpel in an over-filled sharps container or a poorly located container

Suture needle injuries most often occur when:

- Loading or repositioning it in the needle holder
- Passing the needle hand to hand between team members
- Suturing: using fingers to hold tissue or to guide the needle, sewing toward the surgeon or assistant and holding back other tissues by the surgeon or assistant
- Tying with the needle still attached or left on the operative field
- Before and after using the needle: leaving it on the operative field, dropping it on your own or the assistant's foot, and reaching for suture needles or needles loaded in the needle holder sliding off the drapes
- Placing needles in an over-filled sharps container or a poorly located container

Not surprisingly, almost all of these injuries can be easily avoided and with little expense. For example:

- Use a small Mayo forceps (not fingers) when holding the scalpel blade, when putting it on or taking it off or loading the suture needle. (Alternatively, use disposable scalpels with a permanent blade that cannot be removed.)
- Always use tissue forceps, not fingers, to hold tissue when using a scalpel or suturing.
- Use a "hands-free" technique to pass or transfer sharps (scalpel, needles and sharp-tipped scissors) by establishing a Safe or Neutral Zone in the operative field (see below).
- Always remove sharps from the field immediately after use.
- Make sure that sharps containers are replaced when they are only three-quarters full and place containers as close to where sharps are being used as conveniently possible (i.e., within arm's reach).

The "Hands-Free" Technique for Passing Surgical Instruments

A safer method of passing sharp instruments (scalpels, suture needles and sharp scissors) during surgery, called the "hands-free" technique, has recently been recommended. This technique for sharps is inexpensive, simple to use, and ensures that the surgeon, assistant or scrub nurse **never** touches the same instrument at the same time (Bessinger 1988; Fox 1992).

Instruments passed with the hands-free technique (besides those listed above) include anything sharp enough to puncture a glove (e.g., trocars, sharp-tipped mosquito forceps and loaded needle holders). Using the hands-free technique, the assistant or scrub nurse places a sterile or high-level disinfected kidney basin, or other suitable small container, on the operative field between her/himself and the surgeon. The container is designated as the Safe or Neutral Zone in which sharps are placed before

and immediately after use.[1] For example, the assistant or scrub nurse alerts the surgeon that a sharp instrument has been placed in or on the Safe Zone, with the handle pointing toward the surgeon, by saying "scalpel" or "sharp" while placing it there. The surgeon then picks up the instrument and returns it to the container after use, this time with the handle pointing away from her/him.

Note: To avoid dulling scalpel blades, use a plastic container or place a sterile cloth in a metal container.

Another way to do this is to have the assistant or scrub nurse place the instrument in a container and pass it to the surgeon. The surgeon lifts the instrument out of the container, which is left on the field until the surgeon returns the instrument to it. The assistant or scrub nurse then picks up the container and returns it to the Mayo stand.

DESIGNING SAFER OPERATIONS

Using the least dangerous instrument or device that will effectively accomplish the task, while at the same time minimizing risks to the patient and surgical team, should be a goal of any operation. Simple things, such as a brief pre-op discussion of how sharps will be handled by the surgeon, assistant or scrub nurse, can be very helpful. Better still is for the surgical team to review how to make each step in the operation safer, from securing the towel drapes around the proposed incision with nonperforating towel clips to using blunt-tipped needles for closure of all layers except the skin (CDC 1997; Dauleh et al. 1994). Other examples of instruments or devices that protect the surgical team without sacrificing patient safety or staff performance are shown in **Table 7-1**.

In addition, the use of hand-held **straight suture needles** to close skin incisions is especially dangerous, with a reported injury rate of 17%, much higher than with curved needles carried in a needle holder (Davis 2001b). Anesthesiologists, radiologists and others who close small incisions after placement of vascular catheters or cut-downs should be made aware of this hazard.

The risk associated with assisting or being the scrub nurse in surgery may be reduced by anticipating (preferably knowing) the needs of the surgeon for each step of the operation in advance. Where procedures are short (30 minutes or less) and/or the surgical steps are straightforward such as a D&C or cesarean section, this can be accomplished by developing checklists that lay out each step (or task) in the operation or procedure in the sequence in which they usually will be performed (i.e., from skin incision to closure). Reviewing the checklist with the surgical team just before starting the case and pointing out where deviations may be necessary will make the planned surgery go more smoothly and with less

[1] Various items, such as basins, mats or trays, including part of a sterile instrument stand or a designated area on the operative field, have been used as the Safe Zone.

risk of injury. An additional advantage of this review is that it can help protect patients from injury or increased blood loss.

Table 7-1. Reducing the Risk of Exposure

FUNCTION	SAFER	LESS SAFE	LEAST SAFE[1]
Skin incision	cautery	disposable scalpel	scalpel with removable blade
Cutting	scissors, blunt tip or cautery probe	scissors, sharp tip	scalpel
Hemostasis	blunt suture needles staples or cautery	sharp suture needles	wire sutures
Sponging with gauze while using a scalpel	surgeon does sponging; assistant only retracts	assistant sponges but only by request	assistant sponges spontaneously (no communication)
Retraction	blunt retractor	sharp retractor	fingers or hands
Sharps transfer	Neutral Zone	hand-to-hand (communication)	hand to hand (no communication)
Surgical gloves	double gloving	single pair of gloves or double gloving with reprocessed gloves	single pair of reprocessed gloves,
Closing peritoneum (small, 2–3 cm incision)	do not close	purse-string closure using tissue forceps to grasp needle	purse-string closure using fingers to grasp needle

[1] Should be avoided if at all possible.

Blunt Needles for Suturing

The range of "bluntness" in commercially available blunt-tipped needles varies from minimal (no extra effort needed to use them) to very blunt (does not penetrate tissue such as fascia and requires conscious effort). **Minimally blunt needles** can be used for closure of all layers from fascia to skin. **Intermediate blunt needles** require some additional conscious effort to close fascia, but are safer to use. **Very blunt needles** are seldom used except when operating deep in the pelvis where the needle absolutely must be retrieved with fingers. The technique for using blunt needles is as follows:

STEP 1: Use a strong needle holder and lock it fully.

STEP 2: Position the needle in the mid-curve, rather than three-quarters of the way back to prevent slippage or bending the needle. (This usually is not necessary when using minimally blunt needles.)

STEP 3: Grasp and hold the tissue to be sutured with a tissue forceps to make it easier for the needle to go through the tissue being sutured.

In general, the blunter the tip, the more important it is to follow these three steps.

Double Gloving

The transmission of HBV and HCV from surgeon to patient and vice versa has occurred in the absence of breaks in technique and with apparently intact gloves (Davis 2001c). Even the best quality, new latex rubber surgical gloves may leak up to 4% of the time.[2] Moreover, latex gloves, especially when exposed to fat in wounds, gradually become weaker and lose their integrity.

Although double gloving is of little benefit in preventing blood exposure if needlesticks or other injuries occur, it may decrease the risk of blood-hand contact. For example, one recent study showed that surgeons wearing single gloves had a blood-hand contact rate of 14% while surgeons wearing double gloves had only a rate of 5% (Tokars et al 1995; Tokars et al 1992). Based on this study, the following are reasonable guidelines for when to **double glove**:

- The procedure involves coming in contact with large amounts of blood or other body fluids (e.g., vaginal deliveries and cesarean sections).
- Orthopedic procedures in which sharp bone fragments, wire sutures and other sharps are likely to be encountered.
- Surgical gloves are reused. (The possibility of inapparent holes or perforations in any type of reprocessed glove is higher than with new gloves.)

In general, for surgical procedures that are short (30 minutes or less) and involve minimal exposure to blood or mucous secretions (e.g., laparoscopy or minilaparotomy), double gloving is probably not necessary. Whether or not the surgeon, assistant or nurse should double glove should be considered carefully, especially where gloves are reused and in areas where the risk of contracting bloodborne pathogens, such as HIV, is high (>5% prevalence).

Elbow-length Gloves for Obstetrical Procedures

Blood contact with the skin and mucous membranes of providers occurs in 25% of vaginal deliveries and 35% of cesarean sections (Davis 2001d). In addition, large volumes of amniotic fluid contaminated with blood are routine in obstetrics. For skilled birth attendants doing home deliveries, wearing clean examination gloves and avoiding contact with the vaginal area as much as possible is recommended, especially after the membranes have ruptured. Also, changing gloves and washing hands if gloved hands become heavily contaminated with blood or amniotic fluid can minimize the risk of exposure.

[2] The "acceptable" leak rate for new surgical and examination gloves designated by regulatory agencies is up to 4% (Davis 2001c).

Where the hand and forearm need to be inserted into the vagina (manual removal of a retained placenta) or deep into the uterus to deliver the infant's head (cesarean section), elbow-length, so-called "gauntlet" gloves, help protect the provider from significant blood and amniotic fluid contamination. Moreover, by wearing gauntlet gloves, the mother will be protected as well.

If gauntlet gloves are not available, an inexpensive, effective alternative can be easily made from previously used surgical gloves that have been decontaminated, cleaned and dried.[3] The steps for making them are:

STEP 1: Cut the four fingers completely off each glove just below where all the fingers join the glove (**Figure 7-1**).

STEP 2: Sterilize or high-level disinfect 2–3 pairs of cut-off (fingerless) gloves according to the recommended process for each method (**Appendix C**) and store the gloves after final processing in a sterile or high-level disinfected container until needed.

Figure 7-1. Creating Gauntlet Gloves from Previously Used Surgical Gloves

Note: If the need for protection of the forearm(s) occurs **during** a procedure (e.g., removal of a retained placenta), first remove the surgical glove from one or both hands using the technique described in **Chapter 3**. Next, put on a fingerless sterile or high-level disinfected glove(s) and pull up onto the forearm(s). Finally, put a new sterile or high-level disinfected surgical glove on one or both hands.

If it is anticipated that the forearms need to be protected **before** starting the procedure (e.g., cesarean section with presenting part deep in the pelvis), the steps are:

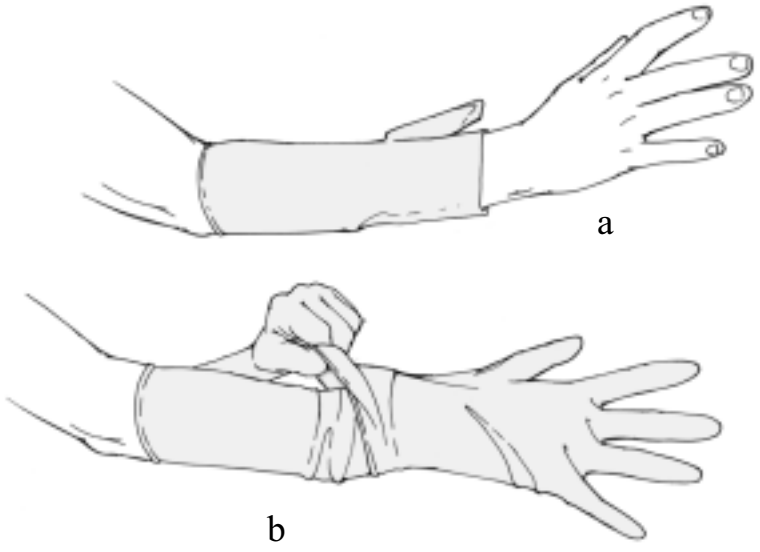
STEP 1: Perform surgical handscrub, including the forearms up to the elbows, as detailed in **Chapter 3** using an alcohol-based antiseptic agent.

STEP 2: Put fingerless sterile or high-level disinfected gloves on both hands and pull up onto the forearm(s) (as shown in **Figure 7-2a**).

STEP 3: Put intact sterile or high-level disinfected surgical gloves on both hands so that the distal (lower) end of the fingerless glove is completely covered (**Figure 7-2b**).

[3] Latex rubber surgical gloves are preferred over examination gloves or even nitrile surgical gloves because they have longer cuffs, are more elastic, fit tighter on the forearm and are more durable.

Figure 7-2a and b. Putting on Fingerless and Surgical Gloves



SAFE HANDLING OF HYPODERMIC NEEDLES AND SYRINGES

In the operating room, scalpels and suture needles are the leading source of penetrating injuries. Hypodermic (hollow bore) needles, however, cause the most injuries to health workers at all levels. Consider:

- Surgeons and assistants are most often stuck by hypodermic needles during procedures.
- Cleaning staff are most often stuck by needles when washing soiled instruments.
- Housekeeping staff are most often stuck by needles when disposing of infectious waste material.

Safety Tips for Using Hypodermic Needles and Syringes

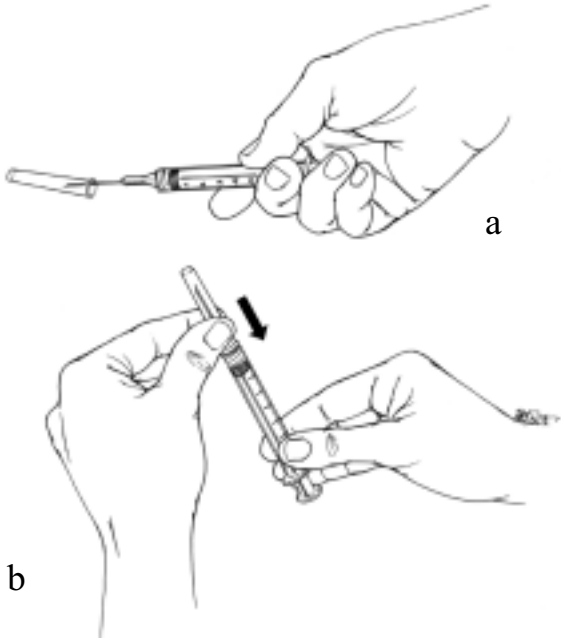
- Use each needle and syringe only once.[4]
- Do **not** disassemble the needle and syringe after use.
- Do **not** recap, bend or break needles prior to disposal.
- Decontaminate the needle and syringe prior to disposal.
- Dispose of the needle and syringe in a puncture-resistant container.

[4] Several studies have documented that unsafe injection practices, such as using the same needle, syringe or both for more than one injection or improperly processed syringes and needles, are responsible for transmitting HIV, HBV and HCV (Drucker, Alcabes and Marx 200l; Simonsen et al 1999). Therefore, after each use, the assembled needle and syringe should either be decontaminated and placed in a sharps container for disposal, or reprocessed using recommended infection prevention practices (see **Chapter 14** and **Appendix E**).

If the needle must be recapped, use the "one-handed" recap method:

- First, place the needle cap on a firm, flat surface; then remove hand.
- Next, with one hand holding the syringe, use the needle to "scoop" up the cap (**Figure 7-3a**).
- With the cap now covering the needle tip, turn the syringe upright (vertical) so the needle and syringe are pointing toward the ceiling.
- Finally, using the forefinger and thumb of your other hand, grasp the cap just above its open end (**Figure 7-3b**) and push the cap firmly down onto the hub (the place where the needle joins the syringe under the cap).

Figure 7-3a and b. One-Handed Recap Method



Safety Tip for Using a Needle and Syringe for Multiple Injections in the Operating Room

If a hypodermic needle must be used for multiple injections during a surgical procedure, one option for preventing accidents between uses is as follows:

- Roll a sterile towel into a tube shape.
- Stick the needle into the towel between uses.

How to Withdraw Medication from a Sterile Multidose Bottle

Note: Do not leave a needle inserted in the rubber stopper of a multidose bottle. This practice provides a direct route for microorganisms, including HIV, to enter the bottle and contaminate the fluid between each use.

- Wipe the top of the bottle with a cotton swab soaked in 60–90% alcohol or other locally available disinfectant. Allow it to dry.
- If using a new disposable needle and syringe, open the sterile pack.
- If using a sterile or high-level disinfected syringe, remove it from the covered container using dry, sterile or high-level disinfected forceps.
- Attach the needle to the syringe.
- Remove the needle cap and insert the needle tip until it touches the bottom of the bottle.
- After filling the syringe, withdraw both the needle and syringe from the bottle.[5]

How to Withdraw Medication Using an Autodisable Syringe

In seeking to improve injection safety, several years ago WHO recommended that all immunizations be given using autodisable syringes. Since then they have been widely used in both campaign and routine immunization settings. Although there are many types of autodisable syringes, the key feature of all of them is that they only permit the syringe to be filled and emptied once. In 2002, USAID began providing the SoloShot FX™ autodisable syringe for use in giving the injectable contraceptive DMPA (Depo Provera®).

The SoloShot FX syringe is a single-use, disposable syringe with a metal clip that locks the plunger after a single use (i.e., it can not be pulled back a second time). The syringe is packaged with a detachable needle, which cannot be attached to any other type of syringes, in a sterile package.

Although autodisable syringes and needles are similar to conventional ones, most health workers will require practice in learning to correctly fill them to avoid wasting medication, syringes and needles (i.e., if air is drawn up into the syringe instead of the prescribed amount of medication, the syringe cannot be refilled). Moreover, it is anticipated that with time use of autodisable syringes for giving other types of injections will increase; therefore, clinicians need to be familiar with using autodisable syringes.

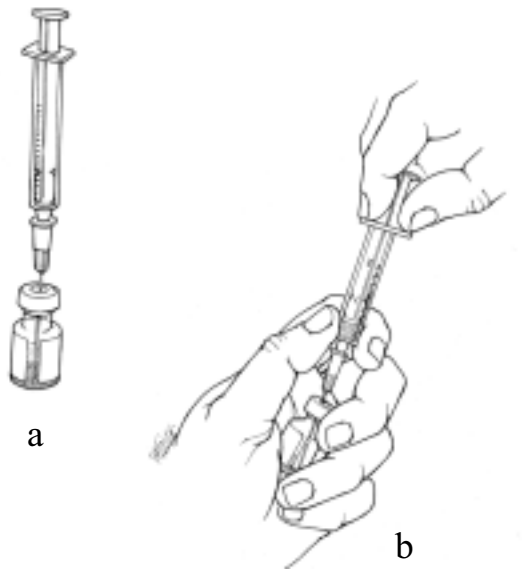
The following instructions are specific for the SoloShot FX syringe and needle:

- Open the sterile pack containing the needle and syringe and attach the needle firmly.
- Remove the needle cap and insert the needle tip until it touches the bottom of the bottle as shown in **Figure 7-4a**. (To avoid drawing air into the syringe, be sure the needle tip stays below the fluid level in the bottle.)

[5] Store opened multidose bottles in a separate, covered container to avoid contamination. Also, mark the date of the first withdrawal. Discard if unused after 30 days or if contaminated at any time.

- While holding the bottle with one hand, slowly pull back on the plunger of the syringe and draw up fluid to just above the "fill line" mark (**Figure 7-4b**).[6]

Figure 7-4a and b. Withdrawing Medication Using an Autodisable Syringe (SoloShot FX™)



- Withdraw the needle and syringe from the bottle and hold the syringe upright (needle pointing to the ceiling) to see if any air is in the syringe.
- If there are air bubbles, slowly push the plunger in, but only until the "fill line" mark is reached.
- Check to be sure the fluid level in the syringe is at or slightly above the "fill line" mark. If it is below the fill line mark, there may not be enough medication to be effective and the injection should not be administered. In this situation, either inject the medication back into the single dose bottle and draw up the medication again using a new autodisable syringe and needle, or discard the partially filled syringe and use a new bottle and autodisable syringe and needle.

SHARPS CONTAINERS: DOs AND DON'Ts

Sharps containers are a key component in minimizing injuries from disposable sharps—such as hypodermic needles, scalpels and suture needles—that are used at all levels of the healthcare system. Other operating room-specific sharps that require similar disposal include: surgical drain trocars, needle point cautery tips, wire sutures, orthopedic drill bits and a range of hollow injection needles used by radiologists and

[6] For the SoloShot FX syringes used with DMPA, the "fill line" mark is at 1 mL.

anesthesiologists for various medical invasive procedures. Disposal of these items after their use requires careful planning and action on the part of the healthcare team to avoid injury to the housekeeping and maintenance staff that ultimately will be removing them.

Note: Educating staff on the safe handling of sharps reduces the risk of injury (Managan et al 2001).

In the US and other developed countries, a whole industry has grown up to meet the increasing demand for sharps containers. Today, sharps containers of all sizes and shapes are available, either disposable or reusable. Most manufactured containers are designed to be wall mounted or attached to a surface and come with special mounting brackets. A few, however, are still designed to be freestanding (ECRI 1993). In most developing countries, these manufactured items are a luxury. As a result, health workers throughout the world have cleverly developed sharps containers from readily available "throw away" items, such as metal food containers made of aluminum, tin or heavy plastic (e.g., cooking oil bottles and cans), heavy-duty cardboard boxes and even the used plastic drinking water bottles with caps that litter the streets and countryside. Although some are safer than others, they all provide a no-cost, sustainable source of disposable sharps containers for use in small clinics, polyclinics and district-level hospitals with limited budgets. Rather than discouraging practitioners from using these items in favor of manufactured products, they should be given help in developing better, safer containers from existing materials (e.g., advised on which items are more appropriate to use).

When using sharps containers, either commercial or locally produced, here are some DOs and DON'Ts to consider:

- **DO** put sharps containers as close to the point of use as possible and practical, ideally within arm's reach. Also, they should be easy to see, recognize and use.
- **DO** attach containers to walls or other surfaces if at all possible.
- **DO** mark them clearly so that people will not unknowingly use them as a garbage container or for discarding cigarettes.
- **DO** place them at a convenient height so staff can use and replace them easily.
- **DO** mark the fill line at the three quarters full level.
- **DON'T** shake a container to settle its contents and make room for more sharps.
- **DON'T** place containers in high traffic areas (corridors outside patient rooms or procedure rooms) where people could bump into them or be stuck by someone carrying sharps to be disposed of.
- **DON'T** place containers on the floor or anywhere they could be knocked over or easily reached by a child.
- **DON'T** place containers near light switches, overhead fans or thermostat controls where people might accidentally put their hand into them.

MANAGING EXPOSURE TO BLOOD AND BODY FLUIDS

Healthcare professionals (physicians, nurses and midwives) who work in high-risk areas such as surgical and obstetrical units should know what to do in the event of a possible blood exposure to themselves or another health worker. Preventing accidents (needlesticks) and other blood or body fluid exposures are the primary means of preventing work-related transmission of HIV or HCV. For HBV, however, an effective vaccine has been available for nearly 20 years. Unfortunately, in many countries, even health professionals have not been immunized against this serious bloodborne disease. Although only about 5% of people who contract hepatitis B die from the disease, a high percentage become chronic carriers or are disabled and cannot work because of permanent damage to the liver (cirrhosis). In addition, hepatitis B infection is a necessary precursor for hepatitis D (HDV) and primary liver cancer.[7] Being vaccinated protects not only the individual, but also fellow workers, other patients and the individual's family.

Hepatitis B Post-Exposure Guidelines

Several studies have demonstrated that, in susceptible persons (i.e., negative hepatitis B surface antigen [HBsAG] test and no history of receiving immune serum globulin), giving hepatitis B immune globulin (HBIG) is better than conventional immune serum globulin (ISG) (or by inference doing nothing) in preventing acute hepatitis B and seroconversion (Desmyter et al 1975; Grady and Lee 1975). For example, in the study by Seeff et al (1975), a randomized clinical trial comparing HBIG to ISG, only 1.4% compared to 5.9% of susceptible individuals developed acute hepatitis, and only 5.6% compared to 20.7 % seroconverted. Both results were statistically significant at the $P < 0.01$ level, and the findings persisted for up to 1 year. In this trial, the first dose of HBIG (5 mL intramuscularly) was given within 7 days of exposure; with the second dose approximately 1 month later. Only brief and mild side effects were noted with either HBIG (3%) or ISG (3.2%). Unfortunately, the availability of HBIG is limited in many countries, but if accidental exposure is reported promptly, there may be time to procure the HBIG and still give it within 7 days of exposure. Whether ISG provides any protection is not known.

The suggested steps for managing an injury is as follows[8]:

STEP 1: Treat the exposure site if appropriate (e.g., an open wound or cut).

STEP 2: If tetanus immunization or boosters are indicated (>10 years since immunization), give it.

[7] HDV is an incomplete virus that is unable to replicate (make more virus particles) in humans without binding to HBV (Davis 2001e).

[8] If exposure is limited to contact with blood or body fluids on intact skin (hands), wash affected areas with soap and water as soon as possible. For contact with mucous membranes (eyes, nose or mouth), rinse with clean water at least two times.

STEP 3: Assess the risk of HBV exposure and determine the immune status of the patient (i.e., history of jaundice, hepatitis or previous immunization with hepatitis B vaccine). If status is unknown, continue assessment.

STEP 4: Collect a specimen from the source person (i.e., the carrier or person suspected of being infected) if possible and from the patient for HBsAGg testing. If testing is not possible, base the HBV status of the infected person on clinical history and clinical findings.

STEP 5: Give HBIG (5mL IM) as soon as possible and within 7 days of exposure, and also give the first dose of hepatitis B vaccine, which should be repeated at 1 and 6 months. If active immunization with hepatitis B vaccine is not possible, a second dose of HBIG should be given 1 month later (Chin 2000).

HIV Post-Exposure Prophylaxis Guidelines

The plan for assessing the risk of accidental exposure to HIV is similar to that for HBV. Because there is no vaccine for passive or active immunization against HIV, post-exposure prophylaxis (PEP) is much more complicated; therefore, the decision to recommend it needs to be based on a careful assessment of the injury. For example, although the risk of HIV seroconversion after all types of work-related percutaneous (breaks the skin) exposure is only about 0.3% (Tokars et al 1993), the risk for deep injuries (extends into the muscle), including deep needlesticks, is 15 times greater than for superficial injuries (CDC 1995; Cardo et al 1997).

If the assessment is positive for a high risk of HIV exposure (i.e., deep injury or needlestick), consider giving treatment with antiretroviral agents (zidovudine [ZDV] plus lamivudine [3TC] has been shown to prevent HIV transmission) (CDC 2001).[9] Determining whether or not PEP should be initiated for a potentially HIV-exposed individual is more difficult than for HBV for three reasons. First, treatment should be initiated as soon as possible and at least within hours after exposure to HIV. Second, a physician or other health professional with knowledge and experience in managing patients with HIV should do the assessment of risk. And third, treatment with antiretroviral agents has considerable side effects, even for prophylaxis, and the long-term safety is not known. Whether or not health workers with exposure to HIV are given PEP, they should receive followup counseling, post-exposure testing and a medical evaluation.

Hepatitis C Post-Exposure Guidelines

There is no post-exposure vaccine or drug prophylaxis for hepatitis C (immune globulin is ineffective). Prevention of exposure, therefore, is the only effective strategy for prevention of HCV.

[9] For the most recent information on post-exposure prophylaxis, go to http://www.cdc.gov/ncidod/hip/guide/phspep.htm.

The CDC (1998) has recommended the following guidelines that institutions should consider for followup of health workers exposed to HCV-positive blood or other body fluids:

- Baseline testing of the source patient (if available and a consent form is signed) for anti-HCV antibody (if the test is available).
- Baseline and 6-month followup testing of exposed health worker for anti-HCV antibody and liver function screen.
- If available, treatment of early HCV infection with pegylated interferon alfa before significant liver damage has occurred.[10]

Where possible, all positive anti-HCV results should be confirmed by supplemental, accurate anti-HCV antibody testing.

MAKING THE SURGICAL ENVIRONMENT SAFER

The responsibility for making today's operating rooms safer extends beyond concern for the well-being of the patient to all healthcare staff who together form the surgical team. The approaches to making operations safer outlined in this chapter are simple, practical and have been documented over a 10-year period. The key to success is to apply the principles and practices in an integrated and consistent manner, with daily attention to detail and, above all, with support at all levels of the healthcare system.

REFERENCES

Bessinger CD Jr. 1988. Preventing transmission of human immunodeficiency virus during operations. *Surg Gynecol Obstet* 167(4): 287–289.

Cardo DM et al. 1997. A case-control study of HIV seroconversion in healthcare workers after percutaneous exposure. *N Engl J Med* 337: 1485–1490.

Centers for Disease Control and Prevention (CDC). 2001. Updated U.S. public health service guidelines for the management of occupational exposures to HBV, HCV, and HIV and recommendations for postexposure prophylaxis. *MMWR* 50(No. RR-11): 26–27.

Centers for Disease Control and Prevention (CDC). 1998. Public health service guidelines for the management of health-care worker exposures to HIV and recommendations for post-exposure prophylaxis. *MMWR* 47(RR-7): 1–33.

[10] Adding polyethylene glycol (PEG) to the interferon molecule increases the half-life of the drug, allowing for less frequent dosing (from three to once a week), but the cost of treatment per month is still nearly USD $2000 (Pharmacology Watch 2002).

Centers for Disease Control and Prevention (CDC). 1997. Evaluation of blunt suture needles in preventing percutaneous injuries among health-care workers during gynecological surgical procedures. *MMWR* 46(2): 25–29.

Centers for Disease Control and Prevention (CDC). 1995. Case-control study of HIV seroconversion in healthcare workers after percutaneous exposures to HIV-infected blood—France, United Kingdom and United States, January 1988–August 1994. *MMWR* 44(50): 929–933.

Chin J (ed). 2000. Viral hepatitis B, in *Control of Communicable Diseases Manual*, 17th ed. American Public Health Association (APHA): Washington, DC, pp 247–251.

Dauleh MI et al. 1994. Needle prick injury to the surgeon—do we need sharp needles? *J R Coll Surg Edinb* 39(5): 310–311.

Davis MS. 2001a. *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta.

Davis MS. 2001b. Blunt alternatives to sharps, in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta, pp 53–63.

Davis MS. 2001c. Choices of effective personal protective equipment, in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta, pp 39–48.

Davis MS. 2001d. Obstetrical procedures, in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta, pp 85–92.

Davis MS. 2001e. Bloodborne pathogens and occupational risk, in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta, pp 1–8.

Desmyter J et al. 1975. Hepatitis-B immunoglobulin in prevention of HBS antigenemia in hemodialysis patients. *Lancet* 2(7931): 376–379.

Drucker EM, PG Alcabes and PA Marx. 2001. The injection century: Consequences of massive unsterile injecting for the emergence of human pathogens. *Lancet* 358: 1989–1992.

ECRI. 1993. Some dos and don'ts for installing and handling sharps containers, in *Technology for Critical Care Nurses*. Plymouth Meeting, PA and from *Nursing96* (February): 51.

Fox V. 1992. Passing surgical instruments, sharps without injury. *AORN J* 55(1): 264.

Grady GF and VA Lee. 1975. Hepatitis B immune globulin—prevention of hepatitis from accidental exposure among medical workers. *N Engl J Med* 293(21): 1067–1070.

Manangan LP. 2001. Infection control dogma: Top 10 suspects. *Infect Control Hosp Epidemiol* 22(4): 243-247.

Pharmacology Watch. 2002. Pegasys approved to treat hepatitis C. December 1.

Seeff LB et al. 1975. Type B hepatitis after needle-stick exposure: Prevention with hepatitis B immune globulin. *Ann Int Med* 88(3): 285–293.

Simonsen L et al. 1999. Unsafe injections in the developing world and transmission of bloodborne pathogens: A review. *Bull World Health Organ* 77(10): 789–800.

Tokars JI et al. 1995. Skin and mucous membrane contacts with blood during surgical procedures: Risk and prevention. *Infect Control Hosp Epidemiol* 16(12): 703–711.

Tokars JI et al. 1993. Surveillance of HIV infection and zidovudine use among healthcare workers after occupational exposure to HIV-infected blood. *Ann Intern Med* 118(12): 913–919.

Tokars JI et al. 1992. Double gloving cuts blood contacts 70%. *Ob/Gyn News* 26(23): 1.

EIGHT

WASTE MANAGEMENT

KEY CONCEPTS you will learn in this chapter include:

- What the purpose of waste management is
- What methods are used to handle contaminated and noncontaminated waste
- How simple, inexpensive incinerators and burial sites are built and used
- What some of the problems of waste removal are

BACKGROUND

Wastes from hospitals and healthcare facilities may be contaminated (potentially infectious) or noncontaminated. Approximately 85% of the general waste produced by hospitals and clinics is **noncontaminated waste** and poses no infectious risk to persons who handle it. Examples of noncontaminated waste include paper, trash, boxes, bottles, plastic containers and food. They can be disposed of by the usual methods or sent to the local landfill or dumpsite (CDC 1985; Rutala 1993).

Some waste from healthcare facilities, however, is contaminated. If not disposed of properly, **contaminated wastes** may carry microorganisms that can infect the people who come in contact with the waste as well as the community at large. Contaminated wastes include blood, pus, urine, stool and other body fluids, as well as items that come in contact with them, such as used dressings. Wastes from operating rooms (human tissue, blood or blood soaked sponges, gauze or cotton) and laboratories (blood, feces, sputum, urine specimens and microbiological cultures) should be considered contaminated. Soiled medical devices or items that can inflict injury (e.g., used needles and scalpel blades) are capable of spreading bloodborne diseases such as hepatitis B, hepatitis C and AIDS, and are also considered contaminated waste.

Other types of waste that do not contain infectious agents, but are considered hazardous because of the potential harm they can cause to the environment include:

- chemical and pharmaceutical residues (e.g., cans, bottles or boxes containing expired drugs and vaccines, laboratory reagents and disinfectants such as formaldehyde and glutaraldehydes, and organic solvents such as acetone and chloroform);
- cytotoxic waste (e.g., drugs typically used in cancer chemotherapy);

- waste with a high content of heavy metals (e.g., mercury from broken thermometers, blood pressure gauges or dentistry materials, and cadmium from discarded batteries); and
- nonrecyclable and discarded pressurized containers (spray cans), that are hazardous if burned because they can explode.

DEFINITIONS

- **Contaminated**. State of having been actually or potentially in contact with microorganisms. As used in healthcare, the term generally refers to the presence of microorganisms that could be capable of producing disease or infection.
- **Container**. Vessel in which waste is placed for handling, transportation, storage and/or eventual disposal.
- **Disposal**. Intentional burial, deposit, discharge, dumping, placing or release of any waste material into or on air, land or water. Disposal is undertaken without the intention of retrieval.
- **Encapsulation**. Filling a sharps container that is three-quarters full with cement or clay, which, after hardening, can be disposed of safely in a landfill.
- **Hazard**. Intrinsic potential property or ability of any agent, equipment, material or process that can cause harm.

Note: Harm is an injury or damage done to the health of people and/or to the environment.

- **Incineration**. Controlled burning of solid, liquid or gaseous combustible (burnable) wastes to produce gases and residues containing little or no burnable material.
- **Infectious waste**. The part of medical waste that is capable of causing infectious diseases.
- **Municipal waste**. General waste for collection by municipalities (e.g., local city or town authorities) generated mainly by households, commercial activities and street-sweeping.
- **Sanitary landfill**. Engineered method of disposing of solid waste on land in a manner that protects the environment (e.g., by spreading the waste in thin layers, compacting it to the smallest practical volume and then covering it with soil at the end of each working day).
- **Scavenging**. Manual sorting of solid waste at landfills and removal of usable material.
- **Segregation**. Systematic separation of solid waste into designated categories.
- **Sewerage**. System for the collection and transport of sewage, including conduits, pipes and pumping stations.
- **Sharps**. Hypodermic needles, suture needles, scalpel blades, scissors, wire sutures, broken glass or any object that can cause a puncture or cut.

- **Waste management**. All activities, administrative and operational (including transportation activities), involved in the handling, treatment, conditioning, storage and disposal of waste.

WASTE MANAGEMENT

The purpose of waste management is to:

- protect people who handle waste items from accidental injury,
- prevent the spread of infection to healthcare workers who handle the waste,
- prevent the spread of infection to the local community, and
- safely dispose of hazardous materials (toxic chemicals and radioactive compounds).

Open piles of waste should be avoided because they:

- are a risk to those who scavenge and unknowingly reuse contaminated items,
- allow persons to accidentally step on sharp items and injure themselves,
- produce foul odors, and
- attract insects and animals.

Disposal of Contaminated Waste

Proper disposal of contaminated waste may include:

Note: If a sewerage system does not exist, dispose of waste in a deep hole and cover.

- Pouring liquids or wet waste directly into a safe sewerage system.
- Incinerating (burning) items to destroy the item as well as any microorganisms. (This is the best method for disposal of contaminated waste. Burning also reduces the bulk volume of waste and ensures that the items are not scavenged and reused.)[1]
- Burying all contaminated wastes to prevent further handling.

Proper handling of contaminated waste minimizes the spread of infection to healthcare personnel and to the local community. Whenever possible, contaminated waste should be collected and transported to disposal sites in leakproof, covered waste containers.

- Use plastic or galvanized metal containers with tight-fitting covers for contaminated wastes. Many facilities now use colored plastic bags to alert handlers to the contents and to keep the general (noncontaminated) waste separate from contaminated waste.

[1] Burning may release toxic chemicals into the air however.

- Use puncture-resistant sharps containers for all disposable sharps (sharps that will not be reused).
- Place waste containers close to where the waste is generated and where convenient for users (carrying waste from place to place increases the risk of infection for handlers). This is especially important for sharps, which carry the highest risk of injury for health workers and staff.
- Equipment that is used to hold and transport wastes must not be used for any other purpose in the clinic or hospital. (Contaminated waste containers should be marked as such.)
- Wash all waste containers with a disinfectant cleaning solution (0.5% chlorine solution plus soap) and rinse with water regularly.
- When possible, use separate containers for combustible and noncombustible wastes prior to disposal. This step prevents workers from having to handle and separate wastes by hand later.

Note: Clean contaminated waste containers each time they are emptied, and clean those for noncontaminated waste when visibly soiled.

Combustible (burnable) wastes include paper, cardboard and contaminated wastes such as used dressings and gauze.

Noncombustible (nonburnable) wastes include glass and metals.

- Use personal protective equipment (PPE) when handling wastes (e.g., heavy-duty utility gloves and closed protective shoes).
- Wash hands or use a waterless, alcohol-based antiseptic handrub after removing gloves when handling wastes.

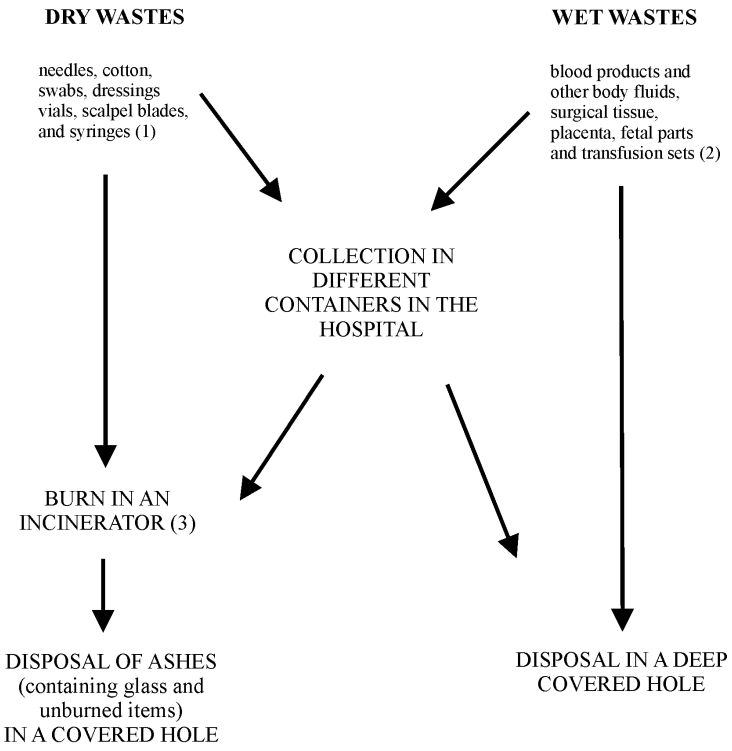
Note: Training staff and having conveniently placed sharps containers available close to where sharps are used will help eliminate the problem of improper disposal.

Because most of the waste from healthcare facilities can be sent to a municipal landfill or dumpsite (the least expensive and easiest way to dispose of waste), it is important to train all healthcare workers, including physicians, to keep contaminated and noncontaminated waste separate. For example, throwing a hypodermic needle into a wastebasket in a patient's room automatically makes that container hazardous for housekeeping staff to handle. And, if discovered, that wastebasket now needs to be handled and disposed of as contaminated waste.

Figure 8-1 is a flow diagram for the separate collection and disposal of wet and dry waste that was first described in Bangladesh (Juncker et al 1994).

In the following sections, specific information and the steps for disposing of sharps, contaminated liquid and solid waste, and hazardous waste items are presented.

Figure 8-1. Flow Diagram: Collection and Disposal of Medical Waste



(1) Small quantities of syringes made of polyethene or polypropene can be incinerated **outside** without producing any environmental health hazard.
(2) Transfusion sets or syringes made of polyvinyl chloride (PVC) should not be incinerated because they release hazardous chemicals.
(3) Built with local materials (e.g., drum or clay single-chamber incinerator; see **Figure 8-3**).

HOW TO DISPOSE OF SHARPS

Disposable sharp items (hypodermic needles, suture needles, razors and scalpel blades) require special handling because they are the items most likely to injure the healthcare workers who handle them as well as people in the community if these items go to the municipal landfill.

Encapsulation

Encapsulation is recommended as the easiest way to safely dispose of sharps. Sharps are collected in puncture-resistant and leakproof containers. When the container is three-quarters full, a material such as cement (mortar), plastic foam or clay is poured into the container until completely filled. After the material has hardened, the container is sealed and may be landfilled, stored or buried. It is also possible to encapsulate chemical or pharmaceutical waste together with sharps (WHO 1999).

Disposal in the Procedure Area

STEP 1: Do not recap needle or disassemble needle and syringe.

STEP 2: After use, to decontaminate the assembled hypodermic needle and syringe, hold the needle tip under the surface of a 0.5% chlorine solution, fill the syringe with solution and push out (flush) three times (if the syringe

Remember: To avoid accidental needlesticks, do not bend, break or recap needles prior to disposal.

and/or needle will be reprocessed, fill the syringe with 0.5% chlorine solution and soak for 10 minutes for decontamination).

See **Appendix E** for more information on handling, processing or disposing of needles and syringes.

Note: The container should be placed at the point of use so that healthcare workers do not have to carry sharp items.

STEP 3: Place assembled needles and syringes to be disposed of in a puncture-resistant sharps container such as a heavy cardboard box, plastic bottle or tin can with lid. The opening in the lid should be large enough that items can be easily dropped through it, but small enough that nothing can be removed from inside. (Old intravenous fluid bottles may also be used, but they can break.)

STEP 4: When the container is three-quarters full, it should be removed from the procedure area for disposal.

Disposing of the Sharps Container

Note: Although suture needles, scalpel blades and other sharp objects may not be completely destroyed by burning, it does make them less likely to be picked up by scavengers.

STEP 1: Wear heavy-duty utility gloves.

STEP 2: When the sharps container is three-quarters full it should be capped, plugged or taped tightly closed. Be sure that no sharp items are sticking out of the container.

STEP 3: Dispose of the sharps container by burning, encapsulating or burying.

STEP 4: Remove utility gloves (wash daily or when visibly soiled, and dry).

STEP 5: Wash hands and dry them with a clean cloth or towel or air dry. (Alternatively, if hands are not visibly soiled, apply 5 mL, about 1 teaspoonful, of an antiseptic handrub and rub the solution vigorously into hands until dry.)

HOW TO DISPOSE OF LIQUID CONTAMINATED WASTES

Liquid contaminated waste (e.g., human tissue, blood, feces, urine and other body fluids) requires special handling, because it may pose an infectious risk to healthcare workers who contact or handle the waste.

Note: Liquid wastes can also be poured into the latrine.

STEP 1: Wear PPE (utility gloves, protective eyewear and plastic apron) when handling and transporting liquid wastes.

STEP 2: Carefully pour wastes down a utility sink drain or into a flushable toilet and rinse the toilet or sink carefully and thoroughly with water to remove residual wastes. **Avoid splashing**.

STEP 3: If a sewage system doesn't exist, dispose of liquids in a deep, covered hole, not into open drains.

STEP 4: Decontaminate specimen containers by placing them in a 0.5% chlorine solution for 10 minutes before washing them.

STEP 5: Remove utility gloves (wash daily or when visibly soiled and dry).

STEP 6: Wash and dry hands or use an antiseptic handrub as described above.

Cholera Epidemic

In case of a cholera epidemic, hospital sewage must also be treated and disinfected. *Vibrio cholerae,* the causative agent of cholera, is easily killed and does not require use of strong disinfectants. Buckets containing stools from patients with acute diarrhea may be disinfected by the addition of chlorine oxide powder or dehydrated lime oxide (WHO 1999).

HOW TO DISPOSE OF SOLID CONTAMINATED WASTES

Remember:
- Never use hands to compress waste into containers.
- Hold plastic bags at the top.
- Keep bags from touching or brushing against the body while lifting or during transport.

Note: If incineration is not available or waste is nonburnable, bury it.

Solid contaminated waste (e.g., surgical specimens, used dressings and other items contaminated with blood and organic materials) may carry microorganisms.

STEP 1: Wear heavy-duty or utility gloves when handling and transporting solid wastes.

STEP 2: Dispose of solid wastes by placing them in a plastic or galvanized metal container with a tight-fitting cover.

STEP 3: Collect the waste containers on a regular basis and transport the burnable ones to the incinerator or area for burning.

STEP 4: Remove utility gloves (wash daily or when visibly soiled and dry).

STEP 5: Wash and dry hands or use an antiseptic handrub as described above.

Special Situations

- If a patient or family member wants to take home the placenta or body parts for burial, first place them in a plastic bag and then into a rigid container (clay bowl, metal or plastic container) for transport.
- Blood and other cultures and stocks of infectious agents from laboratory work should be sterilized by steam sterilization at the earliest stage (i.e., inside the healthcare facility) prior to disposal if possible.

INCINERATION

Incineration is a high-temperature process that reduces the volume and weight of waste. This process is usually selected to treat waste that can not be recycled, reused or disposed of in a sanitary landfill or dumpsite.

Types of Incinerators

Incinerators can range from extremely sophisticated, high-temperature ones to very basic units that operate at much lower temperatures. All types of incinerators, if operated properly, eliminate microorganisms from waste and reduce the waste to ashes.

Four basic types of incinerators are used for treating waste:

1. Double-chamber, high-temperature incinerators are designed to burn infectious waste.
2. Single-chamber, high-temperature incinerators are less expensive and are used when double-chamber incinerators are not affordable.
3. Rotary kilns operate at high temperatures and are used for destroying cytotoxic substances and heat-resistant chemicals.
4. Drum or brick (clay) incinerators operate at lower temperatures and are less effective, but can be made locally using readily available materials.

Note: In this chapter, only inexpensive drum or brick incinerators will be discussed.

Types of Waste That Should Not Be Incinerated

- Pressurized gas containers (aerosol cans)
- Large amounts of reactive chemical waste
- Silver salts and photographic or radiographic wastes
- Plastic containing polyvinyl chloride (blood bags, IV tubing or disposable syringes)
- Waste with high mercury or cadmium content, such as broken thermometers, used batteries and lead-lined wooden panels

Adapted from: WHO 1999.

Open burning is not recommended because it is dangerous, unsightly and the wind will scatter the waste. If open burning must be done, burn in a small, designated area, transport waste to the site just before burning and remain with the fire until it is out.

For healthcare facilities with limited resources and where high-temperature incinerators are not affordable, waste may be incinerated in a drum incinerator. A drum incinerator is the simplest form of single-chamber incinerator. It can be made inexpensively and is better than open burning.

How to Build and Use a Simple Drum Incinerator for Waste Disposal[2]

STEP 1: Where possible, select a site downwind from the clinic.

STEP 2: Build a simple incinerator using local materials (mud or stone) or a used oil drum (e.g., a 55-gallon drum). The size depends on the amount of daily waste collected (**Figure 8-2**).

STEP 3: Make sure the incinerator has:

- Sufficient air inlets underneath for good combustion
- Loosely placed fire bars to allow for expansion
- An adequate opening for adding fresh refuse and for removal of ashes

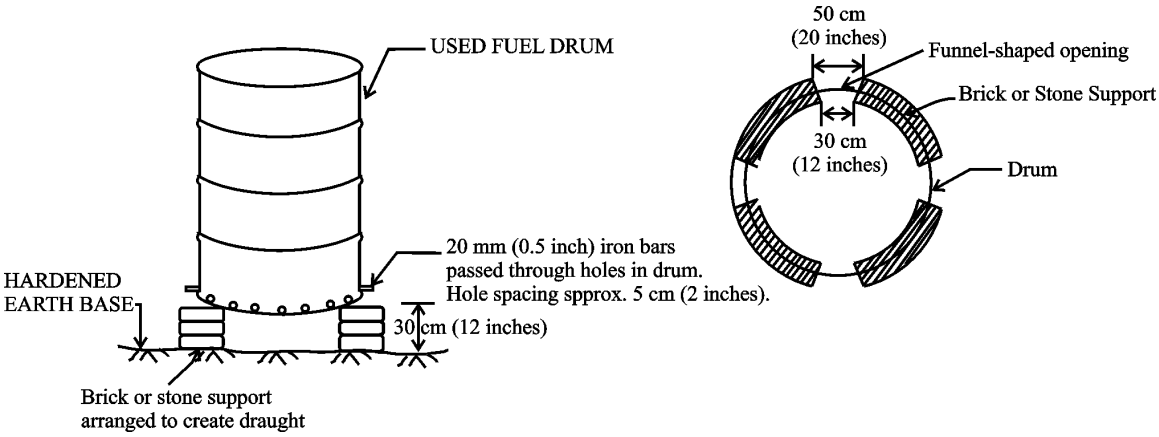
[2] *Adapted from*: SEARO/WHO 1988.

- A long enough chimney to allow for a good draft and evacuation of smoke

STEP 4: Place the drum on hardened earth or a concrete base.

STEP 5: Burn all combustible waste, such as paper and cardboard, as well as used dressings and other contaminated wastes. If the waste or refuse is wet, add kerosene so that a hot fire burns all the waste. Ash from incinerated material can be treated as noncontaminated waste.

Figure 8-2. Design for a Simple Oil Drum Incinerator



Source: SEARO/WHO 1988.

Figure 8-3. Single-Chamber Clay Incinerator

Source: Juncker et al 1994.

BURYING WASTE

Note: Only contaminated and hazardous waste needs to be buried.

In healthcare facilities with limited resources, safe burial of wastes on or near the facility may be the only option available for waste disposal. To limit health risks and environmental pollution, some basic rules are:

- Access to the disposal site should be restricted. (Build a fence around the site to keep animals and children away.)
- The burial site should be lined with a material of low permeability (e.g., clay), if available.
- Select a site at least 50 meters (55 feet) away from any water source to prevent contamination of the water table.
- The site should have proper drainage, be located downhill from any wells, free of standing water and not in an area that floods.

Remember: Large quantities (over 1 kg) of chemical (liquid) wastes should not be buried at the same time; burial should be spread over several days.

Safe on-site burial is practical for only limited periods of time (1–2 years), and for relatively small quantities of waste. During the interval, staff should continue to look for a better, permanent method for waste disposal.

How to Make and Use a Small Burial Site for Waste Disposal[3] (Figure 8-4)

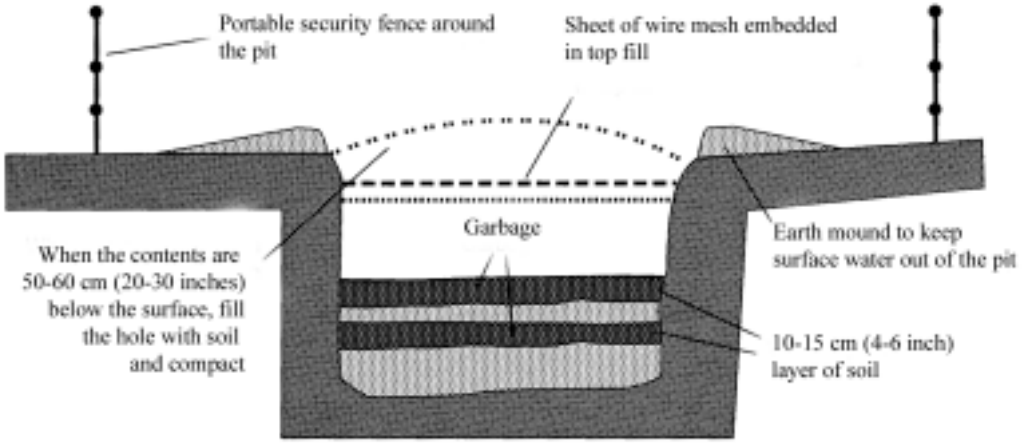
STEP 1: Find an appropriate location (see above).

STEP 2: Dig a pit 1 meter (3 feet) square and 2 meters (6 feet) deep. The bottom of the pit should be 2 meters (6 feet) above the water table.[4]

STEP 3: Dispose of the contaminated waste in the pit and cover the waste with 10–15 cm (4–6 inches) of dirt each day. The final layer of dirt should be 50–60 cm (20–24 inches) and compacted to prevent odors and attraction of insects, and to keep animals from digging up the buried waste.

Depending on the volume of waste, this pit should last 30 to 60 days.

Figure 8-4. Plan for a Small Burial Pit



Adapted from: WHO 1999.

[3] *Adapted from*: SEARO/WHO 1988.
[4] Burial can be used as a method of waste disposal only where the water table is more than 12 feet below the surface.

HOW TO DISPOSE OF HAZARDOUS WASTE[5]

Chemical Waste

Chemical waste includes residues of chemicals in their packaging, outdated or decomposed chemicals, or chemicals that are no longer required. Small **quantities** of chemical waste are generally collected in containers with infectious waste, and are either incinerated, encapsulated or buried. **Large quantities** should **not** be collected with infectious waste. Because there is no safe and inexpensive method for their disposal, the treatment options are the following:

Remember:
- Chemical waste of different types should never be mixed.
- Chemical waste should not be disposed of in sewer systems.

- Incineration at a high temperature is the best option for the disposal of chemical waste.
- If this is not possible, return the chemical waste to the original supplier.

Because either method is expensive and may be impractical, it is important to keep chemical waste to a minimum.

How to Dispose of Used Chemical Containers

- Rinse glass containers thoroughly with water. Glass containers may be washed with soap, rinsed and reused.
- For plastic containers that contained toxic substances such as glutaraldehyde (e.g., Cidex®) or formaldehyde, rinse three times with water and dispose of by burning, encapsulating or burying. Do **not** reuse these containers for other purposes.

Pharmaceutical Waste

Small quantities of pharmaceutical (drugs or medicine) waste are usually placed in containers with infectious waste and disposed of in the same way—either incinerated, encapsulated or safely buried. It should be noted, however, that temperatures reached in a single-chamber drum or brick incinerator may be insufficient to totally destroy the pharmaceuticals; therefore, they can remain hazardous.

Small quantities of pharmaceutical waste, such as outdated drugs (except cytotoxics and antibiotics), may be discharged into the sewer but should not be discharged into natural waters (rivers, lakes, etc.).

Large amounts of pharmaceutical waste may be disposed of by the following methods:

- Cytotoxics and antibiotics may be incinerated with the residues then going to the landfill. (An incinerator, like those used in making cement, that is capable of reaching a combustion temperature of at least 800°C should be used.)[6]

[5] *Adapted from*: WHO 1999.
[6] When incineration is not available, these pharmaceuticals should be encapsulated.

- Water-soluble, relatively mild pharmaceutical mixtures, such as vitamin solutions, cough syrups, intravenous solutions, eye drops, etc., may be diluted with large amounts of water and then discharged to sewers (where sewerage systems exist).
- If all else fails, return pharmaceutical waste to the original supplier if possible.

The following recommendations also should be followed:

- Residues from cytotoxic drugs or other cytotoxic waste should never be mixed with other pharmaceutical waste.
- Cytotoxic waste should never be discharged into natural water sources (rivers, lakes, etc) or landfilled.

Waste with High Content of Heavy Metals

Batteries, thermometers and other items may have a high content of heavy metals, such as mercury or cadmium. Disposal options are as follows:

- Recycling is sometimes available (through cottage industries). This is the best disposal solution when available.
- Encapsulation. If recycling is not feasible, encapsulated waste may be disposed of in a landfill, if available.

This type of waste should not be incinerated because of the toxic metallic vapors emitted, nor should it be buried without encapsulation because it may cause pollution of groundwater. Usually, healthcare facilities have small amounts of this type of waste.

Note: Do **not** touch the droplets with your hands unless wearing examination or utility gloves.

Mercury is a potent neurotoxin, especially during fetal and infant development. When released into water or air, mercury enters the environment, thereby contaminating lakes, rivers and streams. To minimize the risk of mercury pollution, mercury products, such as thermometers and blood pressure equipment, should be replaced with those that do not contain mercury. In case of a spill from a broken thermometer:

- put examination gloves on both hands,
- collect all droplets of mercury with a spoon, and
- place in a small, closed container for disposal or reuse.

Nonrecyclable Aerosol Containers

- Any residual pressure should be released before aerosol containers are buried.
- Pressurized containers should never be burned or incinerated because of the risk of explosion.

In summary, avoid buying or using chemical products that create impossible or very expensive disposal problems wherever possible.

REFERENCES

Centers for Disease Control (CDC). 1985. Recommendations for preventing transmission of infection with human T-lymphotropic virus type III/lymphadenopathy-associated virus in the workplace. *MMWR* 34(45): 681–686; 691–695.

Juncker T et al. 1994. *Clinical disposal at the THC level*. MCH-FP Extension Project (rural). ICDDR: Bangladesh.

Rutala WA. 1993. Disinfection, sterilization and waste disposal, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RD (ed). Williams & Wilkins: Baltimore, MD.

South East Asia Regional Office (SEARO), World Health Organization. 1988. *A Manual on Infection Control in Health Facilities*. SEARO: New Delhi, pp 72–88

World Health Organization (WHO). 1999. *Safe Management of Wastes from Healthcare Activities.* WHO: Geneva.

NINE

OVERVIEW OF RECOMMENDED PROCESSES[1]

KEY CONCEPTS you will learn in this chapter include:

- How soiled instruments, gloves and other reusable items are processed
- How decontamination with 0.5% chlorine solution makes soiled items safer to touch and handle
- When and why each process is used

BACKGROUND

In working to create an infection-free environment, it is important that the rationale for each of the recommended infection prevention processes, and their limitations, be clearly understood by clinic staff at all levels—from healthcare providers to cleaning and maintenance staff.

The basic infection prevention processes recommended to reduce disease transmission from soiled instruments, surgical gloves and other reusable items are **decontamination**, **cleaning** and either **sterilization** or **high-level disinfection (HLD)**. Regardless of the type of operative procedure, the steps in processing surgical instruments and other items are the same, and are illustrated in **Figure 9-1**.

After completing an operation or invasive medical procedure, and while still wearing gloves, the physician or assistant should dispose of contaminated objects (gauze or cotton and other waste items) in a plastic bag or leakproof, covered container. Next, disposable sharps (e.g., scalpel blades and suture needles) should be placed in a sharps container. Finally, all instruments and reusable items such as surgical gloves, syringes and suction cannulae, whether or not they were used in the operation, should be decontaminated by soaking for 10 minutes in a disinfectant (e.g., 0.5% chlorine solution). This step is especially important if these items are to be cleaned by hand (Nyström 1981).

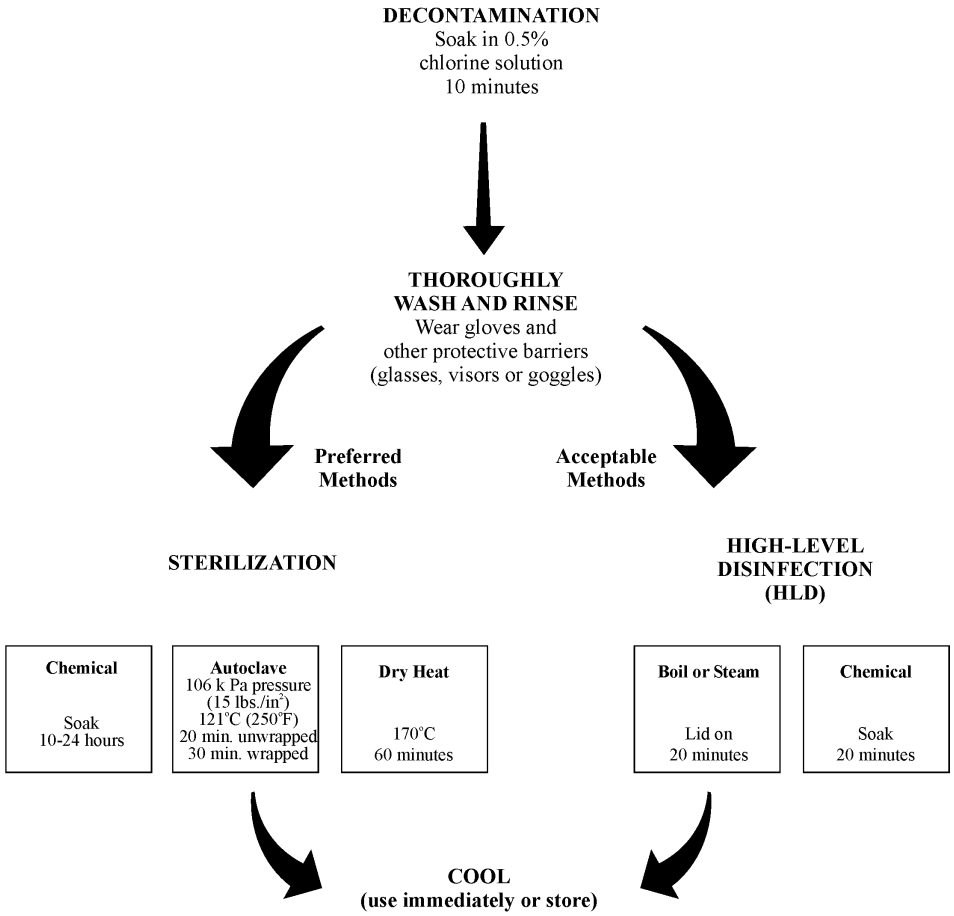
Following decontamination, the instruments and reusable items should be thoroughly **cleaned** with soap and water, completely rinsed and dried. Then, surgical instruments and those items that come in contact with the blood stream or touch normally sterile tissue beneath the skin (critical items) should be **sterilized** to destroy all microorganisms including bacterial endospores.

[1] *Adapted from*: Tietjen, Cronin and McIntosh 1992.

(When sterilization is not feasible or equipment not available, however, HLD by boiling, steaming or soaking in a chemical disinfectant is the only acceptable alternative.) Instruments and other items that touch only mucous membranes or broken skin (semicritical items), however, only need to be high-level disinfected.

Remember: Steaming and boiling, even for prolonged periods, or soaking for 20 minutes in a high-level disinfectant do not destroy endospores reliably. Staff should be aware of the limitations of HLD.

Figure 9-1. Processing Instruments, Surgical Gloves and Other Items



DEFINITIONS

- **Cleaning**. Process that physically removes all visible dust, soil, blood or other body fluids from inanimate objects as well as removing sufficient numbers of microorganisms to reduce risks for those who touch the skin or handle the object. It consists of thoroughly washing with soap or detergent and water, rinsing with clean water and drying.[2]
- **Decontamination**. Process that makes inanimate objects **safer** to be handled by staff **before** cleaning (i.e., inactivates HBV, HBC and HIV

[2] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

and reduces, but does not eliminate, the number of other contaminating microorganisms).

- **High-level disinfection (HLD)**. Process that eliminates **all** microorganisms **except some** bacterial endospores from inanimate objects by boiling, steaming or the use of chemical disinfectants.
- **Sterilization**. Process that eliminates **all** microorganisms (bacteria, viruses, fungi and parasites) **including** bacterial endospores from inanimate objects by high-pressure steam (autoclave), dry heat (oven), chemical sterilants or radiation.

GUIDELINES FOR PROCESSING ITEMS

Each item, whether a soiled metal instruments or pair of surgical gloves, requires special handling and processing in order to:

- minimize the risk of accidental injury or blood or body fluid exposure to cleaning and housekeeping staff; and
- provide a high quality end product (i.e., sterile or high-level disinfected instruments and other items).

Specific guidelines for processing instruments, surgical gloves, equipment and other items used to provide healthcare services are summarized in **Table 9-1**. In this table, the first column lists the item to be processed. The next two columns describe how to decontaminate and clean each item, while in the last two columns the conditions for sterilizing or high-level disinfecting the item, if necessary, are presented.

Additional information and evidence supporting the use of each of these processes is provided in **Chapters 10–12** and **Appendices E–H**. When performed correctly, these processes provide excellent barriers to preventing the spread of infection from medical instruments, surgical gloves and other items to patients and healthcare personnel.

Table 9-1. Guidelines for Processing Instruments, Surgical Gloves and Other Items				
Process	**Decontamination** is the first step in handling used items; it reduces risk of HBV, HCV and HIV viruses.	**Cleaning** removes all visible blood, body fluids and dirt.	**Sterilization** destroys all microorganisms, including endospores.	**High-Level Disinfection** destroys all viruses, bacteria, parasites, fungi and **some** endospores.
INSTRUMENTS OR OTHER ITEMS ITEMS	**DECONTAMINATION**	**CLEANING**	**STERILIZATION[a] OR**	**HIGH-LEVEL DISINFECTION[b]**
Airways (plastic)	Soak in a 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse and wash immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Ambu bags and CPR face masks	Wipe exposed surfaces with gauze pad soaked in 60–90% alcohol or 0.5% chlorine; rinse immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Aprons (heavy plastic or rubber)	Wipe with 0.5% chlorine solution. Rinse with clean water. Between each procedure or each time they are taken off.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.	Not necessary.	Not necessary.
Bed pans, urinals or emesis basins	Not necessary.	Using a brush, wash with disinfectant solution (soap and 0.5% chlorine). Rinse with clean water.	Not necessary.	Not necessary.
Blood pressure cuff	If contaminated with blood or body fluids, wipe with gauze pad or cloth soaked with 0.5% chlorine solution.	If soiled, wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Diaphragms or fitting rings (used for sizing with clients)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Wash with soap and water. Rinse with clean water. Air or towel dry.	Not necessary but can be autoclaved at 121°C (250°F) 106 kPa (15 lbs/in^2) for 20 minutes (unwrapped).	• Steam or boil for 20 minutes. • Chemically high-level disinfect by soaking in 8% formaldehyde, or a 2–4% glutaraldehyde for 20 minutes. Rinse well in water that has been **boiled**.
Exam or operating room tables or other large surface areas (carts and stretchers)	Wipe off with 0.5% chlorine solution.	Wash with soap and water if organic material remains after decontamination.	Not necessary.	Not necessary.

INSTRUMENTS OR OTHER ITEMS ITEMS	DECONTAMINATION	CLEANING	STERILIZATION[a] OR	HIGH-LEVEL DISINFECTION[b]
Footwear (rubber shoes or boots)	Wipe with 0.5% chlorine solution. Rinse with clean water. At the end of the day or when visibly soiled.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.	Not necessary.	Not necessary.
Hypodermic needles and syringes (glass or plastic)	While holding needle under the surface of 0.5% chlorine solution, fill assembled needle and syringe with solution and soak for 10 minutes prior to cleaning. Rinse by flushing three times with clean water.	Disassemble, and then wash with soap and water. Rinse with clean water, air or towel dry syringes (only air dry needles).	**Preferable** (glass only): • Dry heat for 2 hours after reaching 160°C (320°F) (glass syringes only), or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped).	**Acceptable** (glass or plastic): • Steam or boil for 20 minutes. (Chemical HLD is not recommended because chemical residue may remain even after repeated rinsing with boiled water. These residues may interfere with the action of drugs being injected.)
IUDs and inserters (never reuse)	Not appropriate.	Not appropriate.	Not recommended. Most IUDs and inserters come in sterile packages. Discard if package seal is broken.	Not recommended.
Laparoscopes	Wipe exposed surfaces with gauze pad soaked in 60–90% alcohol; rinse immediately.	Disassemble, then using a brush wash with soap and water. Rinse with clean water, towel dry.	Sterilize daily using chemical sterilization. Soak in: • a glutaraldehyde (usually 2%) for 10 hours, or • 8% formaldehyde for 24 hours. Rinse with **sterile** water or water which has been boiled for 20 minutes three times.	Between cases, soak for 20 minutes in: • a glutaraldehyde (usually 2–4%), or • 8% formaldehyde, or • 0.1% chlorine solution with boiled and filtered (if necessary) water. Rinse three times with water that has been **boiled** for 20 minutes.
PPE (caps, masks, covergowns)[d]	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear when handling soiled linen.)	Wash with soap and hot water. Rinse with clean water, air or machine dry. Wrap for reuse.	Not necessary.	Not necessary.
Stethoscopes	Wipe with gauze pad soaked in 60–90% alcohol.	If soiled, wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.

INSTRUMENTS OR OTHER ITEMS ITEMS	DECONTAMINATION	CLEANING	STERILIZATION[a] OR	HIGH-LEVEL DISINFECTION[b]
Storage containers for instruments (metal or plastic)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c]	Wash with soap and water. Rinse with clean water, air or towel dry.	• Dry heat for 1 hour after reaching 170°C (340°F), or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped).	Boil container and lid for 20 minutes. If container is too large: • Fill container with 0.5% chlorine solution and soak for 20 minutes. • Rinse with water that has been **boiled** for 20 minutes and air dry before use.
Suction bulbs (rubber)	Soak in a 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse and wash immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Suction cannulae (plastic) for manual vacuum aspiration (MVA)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Pass soapy water through cannulae three times, removing all particles.	Not recommended. (Heat from autoclaving or dry- heat ovens will damage cannulae.)	Steam or boil for 20 minutes.
Suction catheters (rubber or plastic)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Pass soapy water through catheter three times. Rinse three times with clean water (inside and outside).	Not recommended. (Heat from autoclaving or dry-heat ovens will damage plastic catheters; rubber catheters can be autoclaved.)	• Steam or boil for 20 minutes. (Chemical HLD is not recommended because chemical residue may remain even after repeated rinsing with boiled water.)
Surgical gloves	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Wash with soap and water. Rinse with clean water and check for holes. If to be sterilized, dry inside and out (air or towel dry) and package.	If used for surgery: • Autoclave at 121°C (250°F), and 106 kPa (15 lbs/in^2) for 20 minutes. • Do not use for 24–48 hours.	• Steam for 20 minutes and allow to dry in steamer.
Surgical gowns, linen drapes and wrappers[d]	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear, when handling soiled linen.)	Wash with soap and hot water. Rinse with clean water, air or machine dry.	Autoclave at 120°C/250°F and 106 kPa (15 lbs/in^2) for 30 minutes.	Not practical.

INSTRUMENTS OR OTHER ITEMS ITEMS	DECONTAMINATION	CLEANING	STERILIZATION[a] OR	HIGH-LEVEL DISINFECTION[b]
Surgical instruments (metal)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c]	Using a brush, wash with soap and water. Rinse with clean water. If to be sterilized, air or towel dry and wrap in packs or individually.	**Preferable**: • Dry heat for 1 hour after reaching 170°C (340°F)[e], or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped). **For sharp instruments**: Dry heat for 2 hours after reaching 160°C (320°F).[e]	**Acceptable**: • Steam or boil for 20 minutes. • Chemically high-level disinfect by soaking for 20 minutes. Rinse well with boiled water and air dry before use or storage.
Thermometers (glass)	Not necessary.	Wipe with disinfectant solution (soap and 0.5% chlorine). Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Transfer forceps (chittle) and container (metal)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c] (Reprocess per shift or when contaminated.)	Using a brush, wash with soap and water. Rinse with clean water. If to be sterilized, air or towel dry.	**Preferable**: • Dry heat for 1 hour after reaching 170°C (340°F)[e], or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped).	**Acceptable**: • Steam or boil for 20 minutes. Chemically high-level disinfect by soaking for 20 minutes. Rinse well with boiled water and air dry before use.
Urinary catheters (rubber and straight metal)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c]	Using a brush, wash with soap and water. Rinse three times with clean water (inside and outside).	**Preferable (metal only)**: • Dry heat for 2 hours after reaching 160°C (320°F), or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped).	**Acceptable (rubber or metal)**: • Steam or boil for 20 minutes.
Ventilator tubing or circuits	Not necessary.	Using a brush, wash with soap and water. Rinse with clean water and air dry.	Not possible using an autoclave or dry heat oven.	**Acceptable** • Steam or boil for 20 minutes. • Air dry before use.

[a] If unwrapped, use immediately; if wrapped, reprocess if package becomes damaged or contaminated.
[b] If sterilization (dry-heat or autoclave) is not available, these items can be high-level disinfected either by boiling, steaming or soaking in a chemical disinfectant.
[c] Avoid prolonged exposure (> 20 minutes) to chlorine solution (> 0.5%) to minimize corrosion (rusting) of instruments and deterioration of rubber or cloth products.
[d] Paper or plastic gowns, caps or masks. Place in a plastic bag or leakproof, covered waste container for disposal.
[e] Instruments with cutting edges or needles should **not** be sterilized at temperatures above 160°C to avoid dulling.

REFERENCES

Nyström B. 1981. Disinfection of surgical instruments. *J Hosp Infect* 2(4): 363–368.

Tietjen LG, W Cronin and N McIntosh. 1992. Processing instruments, gloves and other items, in *Infection Prevention Guidelines for Family Planning Service Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 29–43.

TEN

DECONTAMINATION AND CLEANING[1]

KEY CONCEPTS you will learn in this chapter include:

- Why decontamination and cleaning are important initial steps in processing soiled items
- How effective decontamination and cleaning are
- How to prepare dilute bleach solutions for decontamination
- How to decontaminate and clean instruments, surgical gloves and other items

BACKGROUND

Healthcare workers are increasingly at risk of becoming infected with serious bloodborne viruses such as HBV, HCV and HIV. The greatest risk is for staff who:

- perform or assist with surgical procedures (physicians, nurses and midwives);
- process surgical instruments and equipment (staff); and
- perform housekeeping and waste management tasks, including disposal of infectious waste items.

Decontamination and **cleaning** are two highly effective infection prevention measures that can minimize the risk of transmission of these viruses to healthcare workers, especially cleaning and housekeeping staff, when they handle soiled medical instruments, surgical gloves or other items. These measures are also important steps in breaking the infection transmission cycle for patients (see **Chapter 1**). Both processes are easy to do and are inexpensive ways of ensuring that patients and staff are at a lower risk of becoming infected from contaminated instruments and other inanimate objects.

DECONTAMINATION

More than 20 years ago it was shown that decontamination markedly reduces the level of microbial contamination of surgical instruments. For example, in the study by Nyström (1981), 75% of previously soiled instruments had fewer than 10 microorganisms and 98% had fewer than 100 after being decontaminated prior to cleaning. Because of these findings, it was strongly

[1] *Adapted from*: Tietjen, Cronin and McIntosh 1992.

recommended that if instruments and other items are to be cleaned by hand, they first should be decontaminated to minimize the risk of infection following accidental injury to cleaning staff as well as to reduce microbial contamination of their hands.

As presented in **Figure 9-1**, **decontamination** is the first step in processing soiled surgical instruments, surgical gloves and other items. It is important, before cleaning, to decontaminate these items by placing them in a 0.5% chlorine solution for 10 minutes. This step rapidly inactivates HBV, HCV and HIV and makes the items safer to handle by personnel who clean them (AORN 1990; ASHCSP 1986).

Decontamination Products

Chlorine solutions made from sodium hypochlorite generally are the least expensive and the most rapid acting and effective products to use for decontamination, but other agents can also be used such as 70% ethyl or isopropyl alcohol and 0.5–3% phenolic compounds (Crutcher et al 1991).

If no disinfectants are available for decontamination, extreme care must be taken when handling and cleaning sharps (e.g., suture needles, scissors and scalpel blades).

Table 10-1 describes how to make 0.1% and 0.5% chlorine solutions using commercially available liquid bleach products.

Table 10-1. Preparing Dilute Chlorine Solutions from Liquid Bleach (Sodium Hypochlorite Solution) for Decontamination and HLD

TYPE OR BRAND OF BLEACH (BY COUNTRY)	CHLORINE	PARTS WATER TO 1 PART BLEACH[a]	
	% available	0.5%	0.1%[b]
8 °chlorum[c]	2.4%	4	23
JIK (Kenya), Robin Bleach (Nepal)	3.5%	6	34
12 °chlorum	3.6%	6	35
Household bleach (USA, Indonesia), ACE (Turkey), Eau de Javal (France)	5%	9	49
(15 °chlorum), Lejía (Peru)			
Blanquedor, Cloro (Mexico)	6%	11	59
Lavandina (Bolivia)	8%	15	79
Chloros (UK)	10%	19	99
Chloros (UK), Extrait de Javel (France)	15%	29	149
(48 °chlorum[c])			

[a] Read as one part (e.g., cup or glass) concentrated bleach to x parts water (e.g., JIK [0.5% solution]—mix 1 cup bleach with 6 cups water for a total of 7 cups).

[b] Use boiled water when preparing a 0.1% chlorine solution for HLD because tap water contains microscopic organic matter that inactivates chlorine.

[c] In some countries, the concentration of sodium hypochlorite is expressed in chlorometric degrees (°chlorum); one °chlorum is approximately equivalent to 0.3% available chlorine.

Adapted from: WHO 1989.

The formula for making a dilute chlorine solution from **any** concentrated hypochlorite solution is shown in **Figure 10-1**.

Figure 10-1. Formula for Making a Dilute Solution from a Concentrated Solution

- Check concentration (% concentrate) of the chlorine product you are using.
- Determine total parts water needed using **Table 10-1** or the formula below.

$$\textit{Total Parts (TP) water} = \left[\frac{\%\ \textit{Concentrate}}{\%\ \textit{Dilute}}\right] - 1$$

- Mix 1 part concentrated bleach with the total parts water required.

Example: Make a dilute solution (0.5%) from 5% concentrated solution

STEP 1: Calculate TP water: $\left[\frac{5.0\%}{0.5\%}\right] - 1 = 10 - 1 = 9$

STEP 2: Take 1 part concentrated solution and add to 9 parts water.

The approximate amounts (grams) needed to make 0.1% and 0.5% chlorine-releasing solutions from several commercially available chlorine-releasing compounds (dry powders) are listed in **Table 10-2**.

Table 10-2. Preparing Dilute Chlorine Solutions from Dry Powders

AVAILABLE CHLORINE REQUIRED	0.5%	0.1%[b]
Calcium hypochlorite (70% available chlorine)	7.1 g/L[a]	1.4 g/L
Calcium hypochlorite (35% available chlorine)	14.2 g/L	2.8 g/L
NaDCC[c] (60% available chlorine)	8.3 g/L	1.5 g/L
Chloramine **tablets**[d] (1 g of available chlorine per tablet)	20 g/L (20 tablets/liter) [d]	4 g/L (4 tablets/liter) [d]
NaDCC-based **tablets** (1.5 g of available chlorine per tablet)	4 tablets/liter	1 tablet/liter

[a] For dry powders, read x grams per liter (example: Calcium hypochlorite—7.1 grams mixed with 1 liter water).
[b] Use boiled water when preparing a 0.1% chlorine solution for HLD because tap water contains microscopic organic matter that inactivates chlorine.
[c] Sodium dichloroisocyanurate
[d] Chloramine releases chlorine at a slower rate than does hypochlorite. Before using the solution, be sure the tablet is completely dissolved.

Adapted from: World Health Organization (WHO) 1989.

The formula for making a dilute solution from a powder of **any** percent available chlorine is shown in **Figure 10-2**.

Figure 10-2. Formula for Making Chlorine Solutions from Dry Powders

- Check concentration (% concentrate) of the powder you are using.
- Determine grams bleach needed using **Table 10-2** or the formula below.

$$\textit{Grams/Liter} = \left[\frac{\%\ Dilute}{\%\ Concentrate}\right] x\ 1000$$

- Mix measured amount of bleach powder with 1 liter of water.

Example: Make a dilute chlorine-releasing solution (0.5%) from a concentrated powder (35%).

STEP 1: Calculate grams/liter: $\left[\frac{0.5\%}{35\%}\right] x\ 1000 = 14.2\ g / L$

STEP 2: Add 14.2 grams (≈14 g) to 1 liter of water.

WHO (1989) recommends 0.5% chlorine solution for decontaminating instruments and surfaces before cleaning because potable (clean) tap water often is not available for making the solution. In addition, because of the potentially high load of microorganisms and/or other organic material (blood or other body fluids) on soiled items, using a 0.5% solution for decontamination provides a wider margin of safety (Tietjen and McIntosh 1989). For HLD, a 0.1% chlorine solution can be prepared provided boiled and filtered (if necessary) water is used for dilution, and the items have been thoroughly cleaned and rinsed.

Decontamination Tips

- Use a plastic container for decontamination to help prevent:
  - dulling of sharps (e.g., scissors) due to contact with metal containers; and
  - rusting of instruments due to a chemical reaction (electrolysis) that can occur between two different metals (i.e., the instrument and container) when placed in water.
- Do not soak metal instruments that are electroplated (i.e., not 100% stainless steel) even in plain water for more that an hour because rusting will occur.

After decontamination, instruments should be rinsed immediately with cool water to remove visible organic material before being thoroughly cleaned. For example, some healthcare facilities now keep two buckets in the procedure areas or operating rooms, one filled with 0.5% chlorine solution and one with water, so that the instruments can be placed in the water after soaking in the chlorine solution for 10 minutes. Although this will help to

prevent corrosion, even leaving the instruments in plain water for more than 1 hour can lead to rusting.

Hypodermic needles and **syringes** that are to be disposed of should be decontaminated, placed in a puncture-resistant sharps container and, when the container is three-quarters full, burned, encapsulated or buried. If syringes (and needles) are to be reused, however, they should be thoroughly washed and rinsed after decontamination.

Because it is the contaminated needle that **primarily** is responsible for injuries, it is recommended that **only** the syringe, but **not** the needle, be processed for reuse. Doing this is safer than processing both the needle and the syringe (**Appendix E**). Furthermore, as discussed in **Chapter 14**, it reduces costs and creates less contaminated waste than disposing of both.

Remember: The objective of **decontamination** is to protect individuals who handle surgical instruments and other items, which have been in contact with blood or body fluids, from serious diseases.

Large surfaces, such as pelvic examination or operating tables, that may have come in contact with blood and body fluids should be decontaminated. Wiping with a suitable disinfectant such as 0.5% chlorine solution before reuse or when visibly contaminated is an easy, inexpensive way to decontaminate these large surfaces.

Once instruments and other items have been decontaminated, they can safely be further processed. This consists of **cleaning** and finally either **sterilization** or **high-level disinfection**.

CLEANING

Cleaning is important because:

- It is an effective way to reduce the number of microorganisms, especially endospores that cause tetanus, on soiled instruments and equipment.
- Neither sterilization nor high-level disinfection is effective without prior cleaning (Porter 1987).

Note: Many cleaning products contain ammonia, which can interact with bleach and cause the formation of toxic fumes. Check the label of any cleaning product to see if it contains ammonia. (Sometimes you can be alerted to this if you smell ammonia when opening the container.)

A thorough washing with soap and clean water also physically removes organic material such as blood and body fluids.[2] This is important because dried organic material can entrap microorganisms, including endospores, in a residue that protects them against sterilization or disinfection. Organic matter also can partially inactivate some high-level disinfectants, rendering them less effective (AORN 1992; Rutala et al 1998).

Use of soap is important for effective cleaning because water alone will not remove protein, oils and grease (Nyström 1981). The use of hand (bar) or powdered soap is discouraged because the fatty acids in bar soap react with

[2] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

the minerals in hard water leaving a residue or scum (insoluble calcium salt), which is difficult to remove. Using liquid soap, if available, is preferable because it mixes more easily with water than bar or powdered soaps. In addition, liquid soap breaks up and dissolves or suspends grease, oil and other foreign matter in solution so that they can be removed more easily by the cleaning process.

Do not use abrasive cleaners (e.g., Vim® or Comet®) or steel wool because these products can scratch or pit metal or stainless steel. These scratches then become a nesting place for microorganisms, making cleaning more difficult, as well as increasing the chance of corrosion (rusting).

As shown in **Table 10-3**, most microorganisms (up to 80%) in blood and other organic material are removed during the cleaning process. Moreover, following standard cleaning, most nonlumen surgical instruments contained less than 100 colony-forming units (CFU) consisting of relatively nonpathogenic microorganisms (Rutala et al 1998). This study confirms that thorough cleaning is more effective than previously assumed and documents the importance of cleaning in producing a safe product for surgery.

Table 10-3. Effectiveness of Methods for Processing Instruments

METHOD	EFFECTIVENESS (kill or remove microorganisms)	END POINT
Decontamination	Kills HBV and HIV and most microorganisms	10 minute soak
Cleaning (water only)	Up to 50%	Until visibly clean
Cleaning (soap and rinsing with water)	Up to 80%	Until visibly clean
Sterilization	100%	High-pressure steam, dry heat or chemical for recommended time
High-Level Disinfection	95% (does not inactivate some endospores)	Boiling, steaming or chemical for 20 minutes

Once an item is washed it also needs to be rinsed and usually dried. Thorough rinsing with **clean** water removes any soap residue that can interfere with sterilization or HLD.[3] After rinsing, items should be dried, especially if they will be sterilized or high-level disinfected using chemical disinfectants. Water remaining on the items (e.g., surgical instruments) dilutes the solution and may cause the process to fail.

[3] If tap water is contaminated, use boiled or chlorinated water and filter if necessary.

Cleaning Tips

Note: Even when wearing heavy-duty utility gloves, care should be taken to prevent needlesticks or cuts when washing sharps.

- Wear **gloves** while cleaning instruments and equipment. (Thick household or utility gloves work well.) If torn or damaged, they should be discarded; otherwise they should be cleaned and left to dry at the end of the day for use the following day.
- Wear protective eyewear (**plastic visors, face shields, goggles or glasses**) and a plastic apron, if available, while cleaning instruments and equipment to minimize the risk of splashing contaminated fluids into the eyes and onto the body.

To **prevent splashing** keep the items being washed under the surface of the water.

Note: Clean noncritical items such as blood pressure cuffs and stethoscopes with a suitable disinfectant (Weber and Rutala 1993).

- **Instruments** should be washed with a soft brush (an old toothbrush work well) in soapy water. Particular attention should be paid to instruments with teeth, joints or screws where organic material can collect. After cleaning, instruments should be thoroughly rinsed with clean water to remove soap residue that can interfere with chemical disinfectants used for high-level disinfection or sterilization.
- **Syringes (glass or plastic)** when reused should be disassembled only after decontamination and cleaned with soapy water. They then should be thoroughly rinsed (three times) with clean water to remove the soap by expelling the water through the syringe into another container (to prevent contaminating the rinse water), and then dried.
- **Surgical gloves** should be washed in soapy water. Both the inside and outside should be washed and rinsed in clean water until no soap remains. Test gloves for holes by inflating them by hand and holding them under water. (Air bubbles will appear if there are holes.)

Note: If an item cannot be cleaned, it cannot be reused and should be discarded.

- **Rubber or plastic tubing**, such as nasogastic suction tubing for newborns, should be reused only if it can be thoroughly cleaned, rinsed and dried.
- **Oral** or **rectal thermometers** should never be mixed even after cleaning. Keep them in separate containers.
- **Operative endoscopes** (e.g., laparoscopes) must be carefully cleaned because improper cleaning is a common cause of mechanical problems as well as transmission of infections to the next patient (Weber and Rutala 2001). Immediately after use (and before disassembly), wipe all surfaces with a gauze pad soaked with 60–90% alcohol and rinse with cool water. (This step helps protect the person cleaning by inactivating many microorganisms including HIV.) Then completely disassemble the laparoscope and place in warm water containing a nonabrasive soap. Clean all surfaces with a soft brush. Particular attention should be paid to areas where blood and tissue can easily collect—the inner channel of the operating laparoscope, the Falope-Ring® applicator, the trocar and cannula. After cleaning, laparoscopes should be rinsed thoroughly three

times with clean water to remove all soap residue. Excess water should be removed before proceeding with chemical sterilization or high-level disinfection so as not to dilute the chemical solution.

Savlon should not be used for final processing of laparoscopes because it is not a high-level disinfectant and, furthermore, may cloud the lens.

Additional step-by-step instructions on **how to** decontaminate and clean each of the following items are provided elsewhere in this manual:

- linens and other items (see **Chapter 13**)
- rubber gloves (see **Appendix C**)
- surgical instruments (see **Appendix E**)
- needles and syringes (see **Appendix E**)
- laparoscopes (see **Appendix H**)

Finally, if instruments are to be sterilized, they should be packaged or individually wrapped after cleaning. (For instructions on packaging and wrapping instruments for high-pressure steam sterilization or dry heat sterilization, see **Chapter 11** and **Appendix G**.)

REFERENCES

American Society for Hospital Central Service Personnel (ASHCSP) of the American Hospital Association. 1986. *Training Manual for Central Service Technicians*. Enman C (ed). ASHCSP: Chicago, p 155.

Association of Operating Room Nurses (AORN). 1992. Recommended Practices: Disinfection. *AORN J* 56(4): 715–720.

Association of Operating Room Nurses (AORN). 1990. Clinical issues. *AORN J* 52: 613–615.

Crutcher JM, SH Lamm and TA Hall. 1991. Procedures to protect healthcare workers from HIV infection: Category I (healthcare) workers. *Am Ind Hyg Assoc J 5*2(2): A100–103.

Nyström B. 1981. Disinfection of surgical instruments. *J Hosp Infect* 2(4): 363–368.

Porter CW. 1987. Prevention of infection in voluntary surgical contraception. *Biomedical Bulletin* 6(1): 1–7.

Rutala WA et al. 1998. Levels of microbial contamination on surgical instruments. *Am J Infect Control* 26(2): 143–145.

Tietjen LG, W Cronin and N McIntosh. 1992. Decontamination and cleaning procedures, in *Infection Prevention Guidelines for Family Planning Service Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 44–51.

Tietjen LG and N McIntosh. 1989. Infection prevention in family planning facilities. *Outlook* 7: 2–8.

Weber DJ and Rutala WA. 2001. Lessons from outbreaks associated with bronchoscopy. *Infect. Control Hosp. Epidemiol.* 22(7): 403–408.

Weber DJ and Rutala WA. 1993. Environmental issues and nosocomial infections, in *Prevention and Control of Nosocomial Infection*, 2nd ed. Wenzel (ed). Williams & Wilkins: Baltimore, MD, pp 420–450.

World Health Organization (WHO). 1989. *Guidelines on Sterilization and High-Level Disinfection Methods Effective Against Human Immunodeficiency Virus (HIV).* AIDS Series 2. WHO: Geneva.

ELEVEN

STERILIZATION[1]

KEY CONCEPTS you will learn in this chapter include:

- What the common methods of sterilization are
- What the advantages and disadvantages of these methods are
- How to store sterilized items
- What the advantages and disadvantages of other methods of sterilization are

BACKGROUND

Sterilization destroys all microorganisms, including bacterial endospores.

Sterilization should be used for instruments, surgical gloves and other items that come in direct contact with the blood stream or normally sterile tissues (Spaulding 1939). It can be achieved by high-pressure steam (autoclave), dry heat (oven), chemical sterilants (glutaraldehydes or formaldehyde solutions) or physical agents (radiation). Because sterilization is a process, not a single event, all components must be carried out correctly for sterilization to occur.

Effectiveness

To be effective, sterilization requires time, contact, temperature and, with steam sterilization, high pressure. The effectiveness of any method of sterilization is also dependent upon four other factors:

1. **The type of microorganism present.** Some microorganisms are very difficult to kill. Others die easily.
2. **The number of microorganisms present.** It is much easier to kill one organism than many.
3. **The amount and type of organic material that protects the microorganisms.** Blood or tissue remaining on poorly cleaned instruments acts as a shield to microorganisms during the sterilization process.
4. **The number of cracks and crevices on an instrument that might harbor microorganisms.** Microorganisms collect in, and are protected by, scratches, cracks and crevices such as the serrated jaws of tissue forceps.

Note: Although rinsing an item with alcohol and then igniting it with a match (flaming) sometimes is suggested as a method of sterilization, it is not effective!

Finally, without thorough cleaning, which removes any organic matter remaining on the instruments that could protect microorganisms during the

[1] *Adapted from*: Tietjen, Cronin and McIntosh 1992.

sterilization process, sterilization cannot be assured, even with longer sterilization times.

METHODS OF HEAT STERILIZATION

High-pressure, saturated steam using an autoclave, or dry heat using an oven, are the most common and readily available methods used for sterilization.

Remember: When instruments and equipment are sterilized by high-pressure steam (autoclaving), it is essential that steam reach all surfaces. For example, steam sterilizing closed containers will sterilize only the outside of the containers!

High-pressure steam sterilization is an effective method of sterilization but is the most difficult to do correctly (Gruendemann and Mangum 2001). It is generally the method of choice for sterilizing instruments and other items used in healthcare facilities. Where electricity is a problem, instruments can be sterilized in a nonelectric steam sterilizer using kerosene or other fuel as a heat source.

Dry-heat sterilizers (ovens) are good in humid climates but need a continuous supply of electricity, making them impractical in many remote (rural) areas. Furthermore, dry-heat sterilization, which requires use of higher temperatures, can be used only with glass or metal objects—it will melt other substances.

Note: High-speed (flash) prevacuum sterilizers are operated at higher temperatures (134°C/275°F). Sterilizing time for unwrapped instruments by this method is shorter, only taking 4 minutes. Flash sterilization is usually used for individual items.

Standard Conditions for Heat Sterilization

Steam sterilization (Gravity): Temperature should be 121°C (250°F); pressure should be 106 kPa (15 lbs/in^2); 20 minutes for unwrapped items; 30 minutes for wrapped items. Or at a higher temperature of 132°C (270°F), pressure should be 30lbs/in^2; 15 minutes for wrapped items.

Allow all items to dry before removing them from the sterilizer.

Note: Pressure settings (kPa or lbs/in^2) may vary slightly depending on the sterilizer used. When possible, follow manufacturers' recommendations.

Dry heat:

- 170°C (340°F) for 1 hour (total cycle time—placing instruments in oven, heating to 170°C, timing for 1 hour, and then cooling—is from 2–2.5 hours), or
- 160°C (320°F) for 2 hours (total cycle time is from 3–3.5 hours).

Remember:

- Exposure time begins only after the sterilizer has reached the target temperature.
- Do not overload the sterilizer. (Leave at least 7.5 cm [3 inches] between the items and walls of sterilizer.) Overloading alters heat convection and increases the time required to sterilize.

Source: Perkins 1983.

Sterile instruments and other items should be used immediately unless they:

- were wrapped in a double layer of muslin, paper or other appropriate material prior to sterilization; or
- can be stored in a dry, sterile container with a tight-fitting lid.

The material used for wrapping instruments and other items must be porous enough to let steam through but tightly woven enough to protect against dust particles and microorganisms (see **Appendix G** for wrapping and packaging instructions). Wrapped sterile packs should remain sterile until some event causes the package or container to become contaminated. An event can be a tear or worn area in the wrapping, the package becoming wet or anything else that will allow microorganisms to enter the package or container.

Heat Sterilization for Prion Diseases

Prion diseases, such as Creutzfeldt-Jakob disease (CJD), are a group of degenerative brain diseases that have received much attention during the past few years. They occur in animals (dogs, cows and primates) as well as humans and are rapidly fatal once symptoms develop. In humans, CJD remains rare with an incidence of less than 1 per million in the general population (Holman et al 1996). CJD poses a unique infection prevention problem because prions, which are protein-containing infectious agents, can survive recommended heat or high-pressure steam sterilization processes. In addition, chemical disinfectants, including sterilants such as glutaraldehydes and formaldehyde, are not strong enough to eliminate prion infectivity on contaminated instruments and other items. Therefore, surgical instruments and other critical devices contaminated with high-risk tissue (i.e., brain, spinal cord and eye tissue) from patients with known or suspected CJD require special treatment (Rutala and Weber 2001).

Recommendations for caring for patients with known or suspected CJD, as well as handling and processing contaminated instruments and other devices, include the following:

- Because the risk of transmission of prions from patients or noncritical items (e.g., dishes or bedpans) to health workers is low, only Standard Precautions are needed for patients with known or suspected CJD.
- During surgery, put a minimum number of instruments on the operative field and monitor which instruments were used. This reduces the number of instruments requiring special handling and processing.
- After surgery:
    - Avoid handling contaminated instruments.
    - Disposable items and personal protective equipment worn by the surgical team should be placed in a plastic bag and incinerated.

Note: Do not soak contaminated instruments in dilute bleach (0.5% chlorine) solution or wash them.

- Following surgery, noncritical items such as the operating table, Mayo stand and other environmental surfaces can be decontaminated by wiping with a cloth soaked with 0.5% chlorine solution.

- Heat-resistant instruments and other devices should first be decontaminated by placing them in a gravity displacement sterilizer at 121°C (250°F) for 1 hour, or in a prevacuum sterilizer at 134°C (275°F) for 18 minutes.[2]
- After decontamination, clean and sterilize the instruments using the recommended processes (**Chapter 10** and **11**).
- Alternatively, after surgery, soak contaminated instruments and other devices in 1 *N* sodium hydroxide (NaOH) for 1 hour, then clean and sterilize them using recommended processes (Abrutyn, Goldman and Scheckler 1998; Fishman et al 2002).[3, 4]
- Biopsy tissue and surgical specimens should be placed in formalin for 48 hours, then in formic acid for 1 hour and, finally, back into fresh formalin for 48 hours (Abrutyn, Goldman and Scheckler 1998).

STERILIZATION BY STEAM

General Principles

Steam is an effective sterilant for two reasons. **First**, saturated steam is an extremely effective "carrier" of thermal energy. It is many times more effective in conveying this type of energy to the item than is hot (dry) air. In a kitchen, potatoes can be cooked in a few minutes in a steam pressure cooker while cooking may take an hour or more in a hot-air oven, even though the oven is operated at a much higher temperature. Steam, especially under pressure, carries thermal energy to the potatoes very quickly, while hot air does so very slowly. **Second**, steam is an effective sterilant because any resistant, protective outer layer of the microorganisms can be softened by the steam, allowing coagulation (similar to cooking an egg white) of the sensitive inner portions of the microorganism. Certain types of contaminants, however, especially greasy or oily materials, can protect microorganisms against the effects of steam, thus hindering the process of sterilization. This re-emphasizes the need for **thorough cleaning** of objects before sterilization.

Requirements

Steam sterilization requires four conditions: **adequate contact, sufficiently high temperature, correct time** and **sufficient moisture**. Although all are necessary for sterilization to take place, sterilization failures in clinics and hospitals are most often caused by lack of steam contact or failure to attain adequate temperature. All four conditions are discussed, in order of their importance in ensuring complete sterilization by steam, in **Appendix G**. This

[2] Devices and instruments that are not heat-resistant or are difficult to clean should be incinerated.

[3] WHO recommends that contaminated instruments be steam sterilized while they are still soaking in NaOH. This practice, however, is not recommended because of the additional risk of sterilizer damage and exposure of health workers to chemical toxicity. A warning regarding this practice has been posted on the CDC website (http://www.cdc.gov/ncidod/diseases/cjd/cjd_inf_ctrl_qa.hun).

[4] NaOH is caustic and after use must be neutralized before being disposed of by diluting with large amounts of tap water or addition of an acid, such as hydrochloric acid.

appendix also contains detailed instructions for operating steam sterilizers as well as instructions for wrapping and packing the items for sterilization.

Advantages

- Most commonly used, effective method of sterilization.
- Sterilization cycle time is shorter than with dry heat or chemical sterilants.

Disadvantages

- Requires a continuous source of heat (wood fuel, kerosene or electricity).
- Requires equipment (steam sterilizer), which must be expertly maintained to keep it in working condition.
- Requires strict adherence to time, temperature and pressure settings.
- Difficult to produce dry packs because breaks in procedure are common (e.g., not allowing items to dry before removing, especially in hot, humid climates).
- Repeated sterilization cycles can cause pitting and dulling of cutting edges of instruments (i.e., scissors).
- Plastic items cannot withstand high temperatures.

Instructions (Steam Sterilizer)

STEP 1: Decontaminate, clean and dry all instruments and other items to be sterilized.

Note: To help prevent dulling of sharp points and cutting edges, wrap the sharp edges and needle points in gauze before sterilizing. Repair (sharpen) or replace instruments as needed.

STEP 2: All jointed instruments should be in the opened or unlocked position, while instruments composed of more than one part or sliding parts should be disassembled.

STEP 3: Instruments should not be held tightly together by rubber bands or any other means that will prevent steam contact with all surfaces.

STEP 4: Arrange packs in the chamber to allow free circulation and penetration of steam to all surfaces.

STEP 5: When using a steam sterilizer, it is best to wrap clean instruments or other clean items in a double thickness of muslin or newsprint. (Unwrapped instruments must be used immediately after removal from the sterilizer, unless kept in a covered, sterile container.)

Note: Do not allow to boil dry. Steam should always be escaping from the pressure valve.

If using a pressure cooker or kerosene-powered (nonelectric) gravity displacement steam sterilizer, bring the water to a boil and let steam escape from the **pressure valve**; then turn down heat, but keep steam coming out of the pressure valve.

STEP 6: Sterilize at 121°C (250°F) for **30 minutes** for wrapped items, **20 minutes** for unwrapped items; time with a clock.

STEP 7: Wait 20 to 30 minutes (or until the pressure gauge reads zero) to permit the sterilizer to cool sufficiently. Then open the lid or door to allow steam to escape. Allow instrument packs to dry completely before removal, which may take up to 30 minutes. (Wet packs act like a wick drawing in bacteria, viruses and fungi from the environment.) Wrapped instrument packs are considered unacceptable if there are water droplets or visible moisture on the package exterior when they are removed from the steam sterilizer chamber. If using rigid containers (e.g., drums), close the gaskets.

Note: Do not store trays or packs until they reach room temperature. This usually takes about an hour.

STEP 8: To prevent condensation, when removing the packs from the chamber, place sterile trays and packs on a surface padded with paper or fabric.

STEP 9: After sterilizing, items wrapped in cloth or paper are considered sterile as long as the pack remains clean, dry (including no water stains) and intact. Unwrapped items must be used immediately or stored in covered, sterile containers.

Ideally, a steam sterilizer log should be kept, noting time:

- heat begun,
- correct temperature and pressure achieved,
- heat turned down, and
- heat turned off.

Keeping a log can help ensure that the required amount of time will be observed, even when multiple, new or hurried workers are responsible for overseeing sterilization.

STERILIZATION BY DRY HEAT

When available, dry heat is a practical way to sterilize needles and other instruments. A convection oven with an insulated stainless steel chamber and perforated shelving to allow the circulation of hot air is recommended, but dry-heat sterilization can be achieved with a simple oven as long as a thermometer is used to verify the temperature inside the oven.

Effectiveness

Remember: Just as with steam sterilization, thorough cleaning of the object prior to dry heat sterilization is critical. If an instrument is not properly cleaned, sterilization cannot be ensured, regardless of how long the instrument is heated.

Dry-heat sterilization is accomplished by thermal (heat) conduction. Initially, heat is absorbed by the exterior surface of an item and then passed to the next layer. Eventually, the entire object reaches the temperature needed for sterilization. Death of microorganisms occurs with dry heat by a process of slow destruction of protein. Dry-heat sterilization takes longer than steam sterilization, because the moisture in the steam sterilization process significantly speeds up the penetration of heat and shortens the time needed to kill microorganisms.

Advantages

- Effective method, as dry heat by conduction reaches all surfaces of instruments, even for instruments that cannot be disassembled.
- Protective of sharps or instruments with a cutting edge (fewer problems with dulling of cutting edges).
- Leaves no chemical residue.
- Eliminates "wet pack" problems in humid climates.

Disadvantages

- Plastic and rubber items cannot be dry-heat sterilized because temperatures used (160–170°C) are too high for these materials.
- Dry heat penetrates materials slowly and unevenly.
- Requires oven and continuous source of electricity.

Instructions (Dry Heat Oven)

To ensure correct operation, consult specific operating instructions supplied by the oven's manufacturer.

Note: When using dry heat to sterilize instruments wrapped in cloth, be sure that temperature does not exceed 170°C/340°F.

STEP 1: Decontaminate, clean and dry all instruments and other items to be sterilized.

STEP 2: If desired, wrap instruments in aluminum foil or place in a metal container with a tight-fitting, closed lid. Wrapping helps prevent recontamination prior to use. Hypodermic or suture needles should be placed in glass tubes with cotton stoppers.

Note: Use dry heat only for items that can withstand a temperature of 170°C (340°F) (Perkins 1983).

STEP 3: Place loose (unwrapped) instruments in metal containers or on trays in the oven and heat to desired temperature.

STEP 4: **After the desired temperature is reached**, begin timing. The following temperature/time ratios are recommended (APIC 2002):

170°C (340°F)	60 minutes
160°C (320°F)	120 minutes
150°C (300°F)	150 minutes
140°C (285°F)	180 minutes
121°C (250°F)	overnight

Note: Needles and other instruments with cutting edges should be sterilized at lower temperatures (160°C [320°F]), because higher temperatures can destroy the sharpness of cutting edges (Perkins 1983).

Depending on the temperature selected, the total cycle time (preheating, sterilization time and cool down) will range from about 2.5 hours at 170°C to more than 8 hours at 121°C.

STEP 5: After cooling, remove packs and/or metal containers and store. Loose items should be removed with sterile forceps/pickups and used immediately or placed in a sterile container with a tight-fitting lid.

CHEMICAL STERILIZATION

An alternative to high-pressure steam or dry-heat sterilization is chemical sterilization (often called "cold sterilization"). If objects need to be sterilized, but using high-pressure steam or dry-heat sterilization would damage them or equipment is not available (or operational), they can be chemically sterilized.

Note: Chemical sterilization of hypodermic needles and syringes is not recommended, because chemical residues, which may remain even after repeated rinsing with **boiled** water, may interfere with the action of medications being injected.

Some high-level disinfectants will kill endospores after prolonged (10–24 hour) exposure. Common disinfectants that can be used for chemical sterilization include glutaraldehydes and formaldehyde. Sterilization takes place by soaking for at least 10 hours in 2–4% glutaraldehyde solution or at least 24 hours in 8% formaldehyde. Glutaraldehydes, such as Cidex®, are often in short supply and very expensive, but they are the only practical sterilants for some instruments, such as laparoscopes, which cannot be heated. Both glutaraldehydes and formaldehyde require special handling and leave a residue on treated instruments; therefore, rinsing with **sterile** water is essential if the item must be kept sterile. Also, if not rinsed off, this residue can interfere (cause sticking) with the sliding parts of the laparoscope and cloud the lens.

Note: Because boiling and steaming does not reliably inactivate all endospores, rinsing with boiled water can contaminate **sterile** instruments. It is, however, the only acceptable alternative if sterile water is not available.

Although formaldehyde is less expensive than glutaraldehydes, it is also more irritating to the skin, eyes and respiratory tract and is classified as a potential carcinogen (Rutala 1996). When using either glutaraldehydes or formaldehyde, wear gloves to avoid skin contact, wear eyewear to protect from splashes, limit exposure time and use both chemicals only in well-ventilated areas (Clark 1983).

Remember: Do not dilute formaldehyde with chlorinated water, because a dangerous gas (bis-chloromethyl-ether) is produced.

As items are unwrapped after chemical sterilization, they should be transported and stored in a covered, sterile container. (**Table 12-1** provides guidelines for preparing and using glutaraldehydes and formaldehyde solutions.)

Advantages

- Glutaraldehydes and formaldehyde solutions are not readily inactivated by organic materials.
- Both can be used for items that will not tolerate heat sterilization such as laparoscopes.
- Formaldehyde solutions can be used for up to 14 days (replace sooner if cloudy); some glutaraldehydes can be used for up to 28 days. (Check the manufacturers' instructions and see also **Appendix F**).[5]

[5] Although manufacturers provide guidelines for dilution and for how long a solution can be used, many of their claims have not been validated (Gurevich, Yannelli and Cunha 1990). Chemical strip tests can be used to determine the effectiveness of a solution. If these are not available or practical, use the solution only for the minimum recommended time and change it if it is diluted by wet instruments or is visibly cloudy.

Disadvantages

- Glutaraldehydes and formaldehyde are chemicals that cause skin irritation; therefore, all equipment soaked in either solution must be thoroughly rinsed with sterile water after soaking.
- Because glutaraldehydes work best at room temperature, chemical sterilization cannot be assured in cold environments (temperatures less than 20°C/68°F), even with prolonged soaking.
- Glutaraldehydes are expensive.
- Vapors from formaldehyde (classified as a potential carcinogen), and to a lesser degree glutaraldehydes, are irritating to the skin, eyes and respiratory tract. Wear gloves and eyewear, limit exposure time and use both chemicals only in well-ventilated areas.
- Formaldehyde cannot be mixed with chlorine or chlorinated water because a dangerous gas (bis-chloromethyl-ether) is produced.

Instructions (Chemical Sterilization)

STEP 1: Decontaminate, clean and dry all instruments and other items to be sterilized.

STEP 2: Completely submerge items in a clean container filled with the chemical solution and place the lid on the container.

STEP 3: Allow items to soak:

- 10 hours in a glutaraldehyde (check specific product instructions), or
- at least 24 hours in 8% formaldehyde.

Note: Ideally, **three** separate (sequential) rinse containers should be used.

STEP 4: Remove objects from the solution with sterile forceps; rinse all surfaces three times in sterile water and air dry.

STEP 5: Store objects in a sterile container with a tight-fitting lid if they will not be used immediately.

MONITORING STERILIZATION PROCEDURES

Sterilization procedures can be monitored routinely using a combination of biological, chemical and mechanical indicators as parameters.

Biological Indicators

Monitoring the sterilization process with reliable biological indicators at regular intervals is strongly recommended. Measurements should be performed with a biological indicator that employs spores of established resistance in a known population. The biological indicator types and minimum recommended intervals should be:

Remember: Different sterilization processes have different monitoring requirements.

- Steam sterilizers: *Bacillus stearothermophilus*, weekly and as needed
- Dry-heat sterilizers: *Bacillus subtilis*, weekly and as needed

Chemical Indicators

Chemical indicators include indicator tape or labels, which monitor time, temperature and pressure for steam sterilization, and time and temperature for dry-heat sterilization. These indicators should be used on the inside and outside of each package or container.

External indicators are used to verify that items have been exposed to the correct conditions of the sterilization process and that the specific pack has been sterilized. **Internal indicators** are placed inside a pack or container in the area most difficult for the sterilization agent to reach (i.e., the middle of a linen pack). This is the indicator that tells if the item has been sterilized.

Chemical indicators, such as heat sensitive tape or glass vials containing pellets that melt at certain temperatures for a given time, do not guarantee that sterilization has been achieved. They do, however, indicate whether mechanical or procedural problems in the sterilization process have occurred.

Mechanical Indicators

Mechanical indicators for sterilizers provide a visible record of the time, temperature and pressure for that sterilization cycle. This is usually a printout or graph from the sterilizer, or it can be a log of time, temperature and pressure kept by the person responsible for the sterilization process that day.

STORAGE

All sterile items should be stored in an area and manner whereby the packs or containers will be protected from dust, dirt, moisture, animals and insects. This storage area is best located next to or connected to where sterilization occurs, in a separate enclosed area with limited access that is used just to store sterile and clean patient care supplies. In smaller facilities, this area may be just a room off the Central Supply Department or in the operating unit.

- Keep the storage area clean, dry, dust-free and lint-free.
- Control temperature and humidity (approximate temperature 24°C and relative humidity <70%) when possible.
- Packs and containers with sterile (or high-level disinfected) items should be stored 20–25 cm (8–10 inches) off the floor, 45–50 cm (18–20 inches) from the ceiling and 15–20 cm (6–8 inches) from an outside wall.
- Do not use cardboard boxes for storage. Cardboard boxes shed dust and debris and may harbor insects.
- Date and rotate the supplies (first in/first out). This process serves as a reminder, but does not guarantee sterility of the packs.
- Distribute sterile and high-level disinfected items from this area.

Note: Sterile packs will not remain sterile unless properly stored.

Shelf Life

The shelf life of an item (i.e., how long items can be considered sterile) after sterilization is event-related. The item remains sterile until something causes

the package or container to become contaminated—time elapsed since sterilization is not the determining factor. An event can be a tear or worn area in the wrapping, the package becoming wet or anything else that will enable microorganisms to enter the package or container. These events can occur at any time.

Therefore the shelf life of sterilization depends on the following factors:

- Quality of the wrapper or container
- Number of times a package is handled before use
- Number of people who have handled the package
- Whether the package is stored on open or closed shelves
- Condition of storage area (e.g., humidity and cleanliness)
- Use of plastic dust covers and method of sealing (AORN 1992)

Most packages are contaminated as a direct result of frequent or improper handling or storage. To make sure items remain sterile until you need them:

- prevent events that can contaminate sterile packs, and
- protect them by placing them in plastic covers (bags).

Before using any sterile item, look at the package to make sure the wrapper is intact, the seal unbroken and is clean and dry (as well as having no water stains), then you can be reasonably sure it is sterile regardless of when it was sterilized (Gruendemann and Mangum 2001).

In some healthcare facilities where replacement of supplies is limited and the cloth used for wrapping is of poor quality, time as a limiting factor also serves as a safety margin. If plastic covers (bags) are unavailable for the sterilized items, limiting the shelf life to a specific length of time (e.g., 1 month) may be a reasonable decision as long as the pack remains dry and intact.

OTHER STERILIZATION METHODS

Gas Sterilization

The use of **formaldehyde gas** for killing microorganisms was practiced before the turn of the century. One of the first uses of formaldehyde gas was to fumigate rooms, a practice long since shown to be ineffective and unnecessary (Schmidt 1899). There are, however, automatic, low-temperature steam formaldehyde sterilizers that are effective and can be used to process heat-sensitive instruments and plastic items. As mentioned previously, because formaldehyde vapors are irritating to the skin, eyes and respiratory tract, the use of formaldehyde in this form should be limited.

In the United States and several other countries, **ethylene oxide (ETO)** gas is used for sterilization of heat- and moisture-sensitive surgical instruments, such as plastic devices and delicate instruments. Sterilization using ETO, however, is a more complicated (requires a 2-hour exposure time and a long aeration period) and expensive process than either steam or dry-heat sterilization.[6] In addition, it requires sophisticated equipment and skilled staff specially trained for its safe use, making it impractical for use in many countries (Gruendemann and Mangum 2001).

ETO is hazardous to healthcare workers, patients and the environment. Because ETO is moderately toxic when inhaled, regular exposure to low levels (greater than 1 part per million) may produce harmful effects in humans. Moreover, the gas is irritating to the eyes and mucous membranes, and residual ETO on instruments can cause skin injuries and inflammatory reactions in patients. Finally, because ethylene oxide, a toxic product, is classified as a potential carcinogen as well as a mutagen, disposing of it is difficult (Gruendemann and Mangum 2001).

Ultraviolet Light Sterilization

Ultraviolet (UV) light has been used to help disinfect the air for more than 50 years (Morris 1972). For example, UV irradiation can interrupt transmission of airborne infections in enclosed indoor environments where living conditions are poor and people are crowded together. Because UV irradiation has very limited energy, UV light does **not** penetrate dust, mucous or water. Therefore, despite manufacturers' claims, it **cannot** be used to sterilize water. Although in theory intense UV light can be both bactericidal and viricidal, in practice only limited disinfection of instruments can be achieved. This is because the UV rays can kill only those microorganisms that are struck directly by UV light beams. For surfaces that cannot be reached by the UV rays (e.g., inside the barrel of a needle or laparoscope), any microorganisms present will not be killed (Gruendemann and Mangum 2001).

Other disadvantages of UV:

- It requires a reliable source of electricity.
- It is not effective in areas of high relative humidity.
- UV bulbs require frequent cleaning to remain effective.
- Exposure to UV rays can burn the skin and eyes.

As a consequence, UV irradiation is neither a practical nor effective method in most situations (Riley and Nardell 1989).

[6] Items that are sterilized by ETO need to be aerated (to the outside), so that the residual ETO gas can diffuse out of the packages and items. This can take long periods of time leading to complete cycle times of 24 hours or more (Steelman 1992).

Other Chemical Sterilants

- **Paracetic acid (peroxyacetic acid)**. The acid is rapidly effective against all microorganisms, organic matter does not diminish its activity and it decomposes into safe products. When diluted, it is very unstable and must be used with a specially designed automatic sterilizer (APIC 2002). It is usually used for sterilizing different types of endoscopes and other heat-sensitive instruments.
- **Paraformaldehyde**. This solid polymer of formaldehyde may be vaporized by dry heat in an enclosed area to sterilize objects (Taylor, Barbeito and Gremillion 1969). This technique, called "self-sterilization" (Tulis 1973), may be well suited for sterilizing endoscopes and other heat-sensitive instruments.
- **Gas plasma sterilization (hydrogen peroxide based)**. This method can sterilize items in less than 1 hour and has no harmful by products. It does not penetrate well, however, and cannot be used on paper or linen. A specialized sterilizer is required for performing gas plasma sterilization (Taurasi 1997).

REFERENCES

Abrutyn E, DA Goldman and WE Scheckler (eds). 1998. *Saunders Infection Control Reference Service*. WB Saunders Company: Philadelphia, pp 569–570.

American Association of Operating Room Nurses (AORN). 1992. Recommended practices: Sanitation in the surgical practice setting. *AORN J* 56(6): 1089–1095.

Association for Practitioners in Infection Control (APIC). 2002. *APIC Text of Infection Control and Epidemiology on CD-ROM*. APIC: Washington, DC.

Centers for Disease Control and Prevention (CDC). 2002. Questions and answers regarding Creutzfeldt-Jakob disease infection-control practices. Available at: (http//www.cdc.gov/ncidod/diseases/cjd/cjd_inf_ctrl_qa.htm).

Clark RP. 1983. Formaldehyde in pathology departments. *J Clin Pathol* 36(8): 839–846.

Fishman M et al. 2002. Handling of surgical instruments in a presymptomatic familial carrier of Creutzfeldt-Jakob disease. *Am J Infect Control*. 30(5): 303-306.

Gruendemann BJ and SS Mangum. 2001. Ultraviolet irradiation and lights, in *Infection Prevention in Surgical Settings*. WB Saunders Company: Philadelphia, pp 32–35.

Gurevich I, B Yannelli and BA Cunha. 1990. The disinfectant dilemma revisited. *Infect Control Hospit Epidemiol* 11(2): 96–100.

Holman RC et al. 1996. Creutzfeldt-Jakob disease in the United State, 1979–1994: Using national mortality data to access the possible occurrence of variant cases. *Emerg Infect Dis*. 2(4): 333-337.

Morris EJ. 1972. The practical use of ultraviolet radiation for disinfection purposes. *Med Lab Technol* 29(1): 41–47.

Perkins JJ. 1983. *Principles and Methods of Sterilization in Health Sciences,* 2nd ed. Charles C. Thomas Publisher Ltd.: Springfield, IL, pp 95–166; 286–311.

Riley RL and EA Nardell. 1989. Clearing the air, the theory and application of ultraviolet air disinfection. *Am Rev Respir Dis* 139(5): 1286–1294.

Rutala WA and DJ Weber. 2001. Creutzfeldt-Jakob disease: recommendations for disinfection and sterilization. *Clinical Infectious Diseases* 32(9): 1348–1356.

Rutala WA. 1996. APIC guidelines for selection and use of disinfectants. *Am J Infect Control* 24(4): 313–342.

Schmidt A. 1899. *U.S. Patent 630,782: Disinfecting by Means of Formaldehyde*.

Spaulding EH 1939. Studies on chemical sterilization of surgical instruments. *Surg Gyne Obstet* 69: 738–744.

Steelman V. 1992. Ethylene oxide: the importance of aeration. *AORN Journal* 55(3): 773–787.

Taurasi AR. 1997. Ethylene oxide alternatives www.cea.purdue.edu/iahcsmm/28LESSON.HTM. Downloaded 13 Jan 98.

Taylor LA, MS Barbeito and GG Gremillion. 1969. Paraformaldehyde for surface sterilization and decontamination. *Applied Microbiol* 17(4): 614–618.

Tietjen LG, W Cronin and N McIntosh. 1992. Sterilization, in *Infection Prevention Guidelines for Family Planning Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 52–73.

Tulis JJ. 1973. Formaldehyde gas as a sterilant, in *Industrial Sterilization: International Symposium*. Briggs Phillips G and WS Miller (eds). Duke University Press: Durham, NC, pp 209–238.

TWELVE

HIGH-LEVEL DISINFECTION[1]

KEY CONCEPTS you will learn in this chapter include:

- What the methods of high-level disinfection (HLD) are
- How to perform each method of high-level disinfection
- What the advantages and disadvantages of boiling and steaming are
- What the advantages and disadvantages of chemical high-level disinfectants are

BACKGROUND

Although sterilization is the safest and most effective method for the final processing of instruments, often sterilization equipment is either not available or not suitable (Rutala 1996). In these cases, HLD is the only acceptable alternative. The HLD process destroys all microorganisms (including vegetative bacteria, tuberculosis, yeasts and viruses) except some bacterial endospores.

High-level disinfection can be achieved by boiling in water, steaming (moist heat) or soaking instruments in chemical disinfectants. To be effective, all steps in performing each method must be monitored carefully.

EFFECTIVENESS OF MOIST HEAT

Essentially all vegetative forms of bacteria are killed by moist heat at temperatures of 60–75°C within 10 minutes (Salle 1973). Hepatitis B virus, which is one of the most difficult viruses to kill, is inactivated in 10 minutes when heated to 80°C (Kobayashi et al 1984; Russell, Hugo and Ayliffe 1982). In contrast, although many types of spores are killed when boiled at 99.5°C for 15 to 20 minutes (Williams and Zimmerman 1951), *Clostridium tetani* spores are quite heat-resistant and can even survive boiling for up to 90 minutes (Spaulding 1939).

The highest temperature that boiling water or low-pressure steam will reach is 100°C (212°F) at sea level. Because the boiling point of water is 1.1°C lower for each 1,000 feet in altitude, it is best to boil or steam items to be high-level disinfected for a minimum of 20 minutes. This provides a margin of safety for variations in altitudes up to 5,500 meters (18,000 ft), and at the same time eliminates the risk of infection from some, but not all, endospores.

[1] *Adapted* by: Tietjen, Cronin and McIntosh 1992.

Boiling Versus Steaming

Boiling and steaming both use moist heat to kill microorganisms. **Steaming has several distinct advantages over boiling** for the final processing of surgical gloves and other items, such as plastic cannulae and syringes. It is less destructive and, because it uses much less fuel than boiling, it is more cost-effective. For example, only about 1 liter of water is needed to steam gloves or instruments, whereas 4–5 liters are required for boiling. Also, discoloration of instruments from calcium or other heavy metals contained in some tap water does not occur, because the steam contains only pure water molecules. Finally, although boiling and steaming gloves are equally easy to do, drying boiled gloves is not practical because it is difficult to prevent contamination while they are air drying. With steaming, because they remain in the closed steamer pan, gloves are less likely to become contaminated.

The **major disadvantage of steaming** is that if the steamers available locally are small, they are only practical for use with a small number of items (e.g., one set of instruments or 15–20 pairs of surgical gloves) per tray or pan. For steaming to be effective, the bottom pan must contain enough water to continue boiling throughout the steaming process. By contrast, large boiling pots are easier to use for metal instruments and do not have to be monitored the entire time to be sure that the process is being done correctly.

Both boiling and steaming share some advantages and disadvantages over chemical high-level disinfection, which is the only other method of HLD.

Advantages

- Inexpensive procedures.
- Easily taught to healthcare workers.
- Require no special chemicals or dilutions and leave no chemical residue.
- Heat sources (boilers or rice cookers) are commonly available.

Disadvantages

- Length of processing time must be carefully measured (i.e., start timing only after steam begins to escape or water has reached a rolling boil). Once timing starts, no additional items or water can be added.
- Objects cannot be packaged prior to HLD; therefore, there is a greater chance of contamination if items are to be stored.
- Requires a fuel source that may be unreliable.

HIGH-LEVEL DISINFECTION BY BOILING

Boiling in water is an effective, practical way to high-level disinfect instruments and other items. Although boiling instruments in water for 20 minutes will kill all vegetative forms of bacteria, viruses (including HBV, HCV and HIV), yeasts and fungi, boiling will **not** kill all endospores reliably.

Instructions for HLD by Boiling

STEP 1: Decontaminate and clean all instruments and other items to be high-level disinfected.

STEP 2: If possible, completely immerse items in the water.[2] Adjust the water level so that there is at least 2.5 cm (1 inch) of water **above** the instruments. In addition, make sure all bowls and containers to be boiled are full of water. For example, empty bowls that turn bottom side up and float to the surface contain air pockets.

Remember: A gentle rolling boil is sufficient and will prevent instruments or other items from being bounced around and possibly damaged by striking other instruments or the side walls of the boiling pot.

STEP 3: Close lid over pan and bring water to a gentle, **rolling** boil. (Boiling too vigorously wastes fuel, rapidly evaporates the water and may damage delicate [or sharp] instruments or other items.)

STEP 4: Start timer. In the HLD log, note time on the clock and record the time when rolling boil begins.

STEP 5: Boil all items for **20 minutes**.

Boiling Tips

- Always boil for 20 minutes in a pot with a lid.
- Start timing when the water begins to boil.
- Metal instruments should be completely covered with water during boiling.
- Do not add anything to the pot after timing begins.

STEP 6: After boiling for 20 minutes, remove objects with previously high-level disinfected forceps. Never leave boiled instruments in water that has stopped boiling. As the water cools and steam condenses, air and dust particles are drawn down into the container and may contaminate the instruments (Perkins 1983).

STEP 7: Use instruments and other items immediately or, with high-level disinfected forceps or gloves, place objects in a high-level disinfected container with a tight-fitting cover. Once the instruments are dry, if any pooled water remains in the bottom of the container, remove the dry items and place them in another high-level disinfected container that is dry and can be tightly covered.

Protecting the Life of Instruments That Are Frequently Boiled

Lime deposits may form on metal instruments that are frequently boiled. This scale formation, caused by lime salts in the water, is difficult to avoid. By following these steps, however, the problem of lime deposits can be minimized:

[2] A study documented that the interior temperature of a plastic cannula floating on the surface of boiling water reaches a temperature of 96–98°C in less than 1 minute. Therefore, for items that float (e.g., syringes, plastic MVA cannulae or rubber items), it is not necessary that they be fully covered by the water to achieve HLD if the pot is covered with a lid (IPAS 1993).

STEP 1: Boil the water for 10 minutes at the beginning of each day before use. (This precipitates much of the lime salt in the water on to the walls of the boiling pot before objects are added.)

STEP 2: Use the same water throughout the day, adding only enough to keep the surface at least 1 inch above the instruments to be high-level disinfected. (Frequent draining and replacing the water, and boiling too vigorously, increase the risk of lime deposits on instruments.)

STEP 3: Drain and clean the boiler or pot at the end of each day to remove lime deposits.

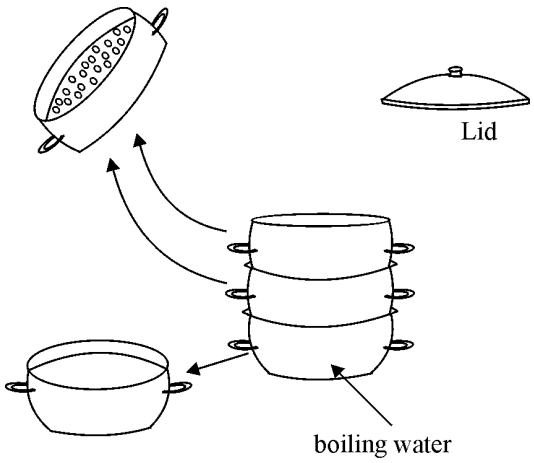
HIGH-LEVEL DISINFECTION BY STEAMING

Steaming surgical gloves has been used as the final step in processing gloves for many years in Indonesia and other parts of Southeast Asia. In 1994, a study by McIntosh et al confirmed the effectiveness of this process.

The steamer used in the study (**Figure 12-1**) consisted of:

- a bottom pan (approximately 31 cm in diameter) for boiling water;
- one, two or three circular pans with multiple 0.5 cm (diameter) holes in their bottoms to permit the passage of steam through them and water back down to the bottom pan; and
- a lid that fits on the top pan.

Figure 12-1. Steamer Used for HLD

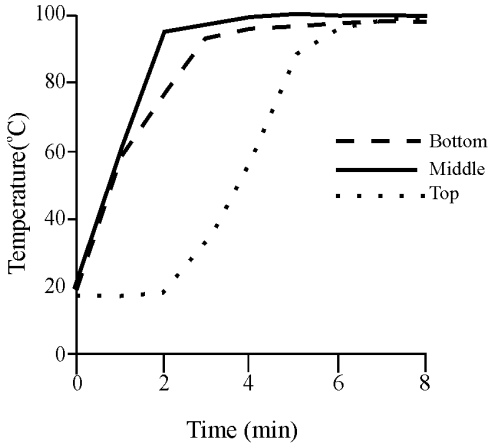


Two types of tests were conducted to determine whether surgical gloves and other items could be high-level disinfected using this process.

In the first set of experiments, a thermocouple was placed inside a glove in each of the three pans and the rate and extent of the temperature change was recorded. As shown in **Figure 12-2**, when 5–15 pairs of surgical gloves were

placed in each of the three pans, the temperature reached 96–98°C in less than 4 minutes in the bottom and middle pans and within 6 minutes in the upper pan. Thereafter, the temperature remained constant throughout the remaining 20 minutes.

Figure 12-2. Temperature Rise in Gloves as a Function of Tray Position



In the second set of experiments, batches of new surgical gloves were contaminated with *Staphylococcus epidermidis*, *Staphylococcus aureus*, *Pseudomonas aeruginosa* and *Candida albicans* as well as *Bacillus subtilis* (heat-sensitive) and *Bacillus stearothermophilus* (heat-resistant) endospores. Next, the gloves were placed in each of the three pans and steamed for 20 minutes. After this, the gloves were removed from the pans and incubated for 24 hours in sterile media and then were plated on blood agar. In all cases (6, 15 and 30 gloves per pan), there was no growth of any microorganisms or *B. subtilis* endospores at 24 hours. As expected, however, only a reduction in the number of *B. stearothermophilus* (heat-resistant) endospores occurred.

Instructions for HLD by Steaming

After instruments and other items have been decontaminated and thoroughly cleaned, they are ready for HLD by steaming. (See **Appendix C** for HLD of surgical gloves by steaming.)

STEP 1: Place instruments, plastic MVA cannulae and other items in one of the steamer pans with holes in its bottom (**Figure 12-1**). To make removal from the pan easier, do not overfill the pan.

STEP 2: Repeat this process until up to three steamer pans have been filled. Stack the filled steamer pans on top of a bottom pan containing water for boiling. A second empty pan without holes should be placed on the counter next to the heat source (see **Step 7**).

Remember: Be sure there is sufficient water in the bottom pan for the **entire** 20 minutes of steaming.

STEP 3: Place a lid on the top pan and bring the water to a full **rolling** boil. (When water only simmers, very little steam is formed and the temperature may not get high enough to kill microorganisms.)

STEP 4: When steam begins to come out between the pans and the lid, start the timer or note the time on a clock and record the time in the HLD log.

STEP 5: Steam items for 20 minutes.

STEP 6: Remove the top steamer pan and put the lid on the pan that was below it (the pan now on top). Gently shake excess water from the pan just removed.

STEP 7: Put the pan just removed onto the empty pan (see **Step 3**). Repeat until all pans are restacked on this empty pan and the top pan is covered with the lid. (This step allows the items to cool and dry without becoming contaminated.)

STEP 8: Allow items to air dry in the steamer pans (1 to 2 hours) before using.

STEP 9: Using a high-level disinfected forceps, transfer the dry items to a dry, high-level disinfected container[3] with a tight-fitting cover. Instruments and other items can also be stored in the stacked and covered steamer pans as long as a bottom pan (no holes) is used.

HIGH-LEVEL DISINFECTION USING CHEMICALS

Although a number of disinfectants are commercially available in most countries, four disinfectants—**chlorine**, **glutaraldehydes**, **formaldehyde** and **peroxide**—are routinely used as high-level disinfectants. (**Table 12-1** provides guidelines for preparing and using these disinfectants.) These chemicals can achieve high-level disinfection if the items being disinfected are thoroughly cleaned before immersion. A high-level disinfectant should be selected for use based on the characteristics of the items to be disinfected, the physical area (i.e., is it well ventilated) and the skills of personnel available to do the procedure.

Note: Chemical HLD of hypodermic needles and syringes is not recommended, because chemical residues, which may remain even after repeated rinsing with **boiled** water, may interfere with the action of medications being injected.

The major **advantages** and **disadvantages** of these high-level disinfectants are:

- **Chlorine solutions** are fast acting, very effective against HBV, HCV and HIV/AIDS, inexpensive and readily available (CDC 1987; WHO 1989). A major disadvantage is that concentrated chlorine solutions (>0.5%) can corrode metals; however, stainless steel and plated instruments can be safely high-level disinfected in 0.1% chlorine solution by soaking in a plastic container for up to 20 minutes. For **HLD**, the 0.1% chlorine solution should be made using **boiled** water, which has been filtered if the tap water is cloudy. Prior to soaking, the items should have been thoroughly cleaned, rinsed and dried.

[3] How to prepare a high-level disinfected container: For small containers, boil water in the covered container for 20 minutes, then pour out the water, which can be used for other purposes, replace the cover and allow container to dry. Alternatively, and for large containers, fill a plastic container with 0.5% chlorine solution and immerse the cover in chlorine solution as well. Soak both for 20 minutes. (The chlorine solution can then be transferred to another container and reused.) Rinse the cover and the inside of the container three times with boiled water and allow to air dry.

Problems from discoloration can be decreased if items are rinsed with boiled water and dried **promptly**.[4] Although chlorine solutions for HLD may deteriorate if left standing uncovered or stored in a clear (transparent) container, fresh solutions for HLD need to be made only if the solution is visibly cloudy.

Note: Using the lower chlorine concentration (0.1%) is just as effective and will extend the useful life of the instruments.

Tables 10-1 and **10-2** describe how to make 0.1% chlorine solutions from commercially available liquid bleach products and dry powders, respectively.

Note: If stored in closed, dark bottles that block light, various concentrations of commercial bleach solutions (1:100 to 1:5) do not lose their efficacy as fast as formerly thought (e.g., 50% to 97% potency at 30 days) with higher concentrations being more stable (Rutala et al 1998).

- **Formaldehyde** (8%), which is inexpensive and readily available, is an effective high-level disinfectant (HLD) but, as mentioned previously, the vapors are very irritating and it is classified as a potential carcinogen. Care must be taken to protect both staff and patients from the fumes when mixing and using formaldehyde solutions. **Do not dilute with chlorinated water as a dangerous gas (bis-chloromethyl-ether) can be produced.** Staff should wear gloves to avoid skin contact, protect eyes from splashes, limit exposure time and use these solutions only in a well-ventilated area.
- **Glutaraldehydes** are less irritating than formaldehyde, but staff and clients still need to be protected from the fumes when mixing and using these solutions. Staff should wear gloves and protective eyewear to avoid skin contact, protect eyes from splashes, limit exposure time and use only in a well-ventilated area.

Remember: Because both glutaraldehydes and formaldehyde (formalin) solutions leave a residue, instruments must be rinsed thoroughly with **boiled** water **three** times after chemical HLD to remove any residue and prevent skin irritation.

- **Hydrogen Peroxide** (H_2O_2), which must be diluted to a 6% solution, often is available locally and is less expensive than other chemical disinfectants. The 3% H_2O_2 solutions used as antiseptics, however, should not be used as a disinfectant. The major disadvantage of peroxide is that it is highly corrosive and should not be used to disinfect copper, aluminum, zinc or brass. Also, because it loses potency rapidly when exposed to heat and light, it should be stored in a cool, dark place. WHO does not recommend using H_2O_2 in hot (tropical) climates because of its instability in the presence of heat and light (WHO 1989).

Advantages and disadvantages of each of these chemical disinfectants are summarized in **Appendix F**.

[4] Discoloration of metal items, which occurs when calcium (not sodium) hypochlorite powders are used, often is confused with corrosion (rusting). Wiping discolored items with a cloth soaked with vinegar (dilute acetic acid) will quickly remove discoloration.

Alcohols and Iodophors

Although alcohols and iodophors are inexpensive and readily available, they are no longer classified as high-level disinfectants. Alcohols do not kill some viruses and are not sporicidal, and *Pseudomonas* species have been shown to multiply in iodophors (Favero 1985; Rutala 1993). These chemicals should be used only when the high-level disinfectants listed above are not available or appropriate.

Key Steps in Chemical High-Level Disinfection

- Decontaminate instruments and other items that may have been contaminated with blood and body fluids, and thoroughly clean and dry them before placing them in the disinfectant solution.
- Completely immerse all items in the high-level disinfectant.
- Soak for 20 minutes.
- Remove items using high-level disinfected or sterile forceps or gloves.
- Rinse well with **boiled** and filtered (if necessary) water three times and air dry.
- Use promptly or store in a dry, high-level disinfected, covered container.

Adapted from: Tietjen and McIntosh 1989.

Storage of Disinfectants

- Chemical disinfectants should be stored in a cool, dark area.
- Never store chemicals in direct sunlight or in excessive heat (e.g., upper shelves in a tin-roofed building).

Disposal of Used Chemical Containers

- **Glass containers** may be washed with soap, rinsed, dried and reused. Alternatively, thoroughly rinse glass containers (at least two times) with water and dispose of by burying.[5]
- **Plastic containers** used for toxic substances such as glutaraldehydes or formaldehyde should be rinsed (at least three times) with water and disposed of by burning or burying.

Disposal of Used Chemicals

Carefully pour wastes down a utility sink drain or into a flushable toilet and rinse or flush with water. Liquid wastes can also be poured into a latrine. **Avoid splashing.** Rinse the toilet or sink carefully and thoroughly with water to remove residual wastes.

[5] To further prevent them from being misused, put a hole in each container before disposal so that water or other liquids cannot be carried in it.

Table 12-1. Preparing and Using Chemical Disinfectants

CHEMICALS FOR STERILIZATION OR HIGH-LEVEL DISINFECTION

Disinfectant (common solution or brand)	Effective Concentration	How to Dilute	Skin Irritant	Eye Irritant	Respiratory Irritant	Corrosive	Leaves Residue	Time Needed for HLD	Time Needed for Sterilization	Activated Shelf Life[a]
Chlorine	0.1%	Dilution procedures vary[b]	Yes (with prolonged contact)	Yes	Yes	Yes[c]	Yes	20 minutes	Do not use	Change every 14 days, sooner if cloudy.
Formaldehyde (35–40%)	8%	1 part 35–40% solution to 4 parts boiled water	Yes	Yes	Yes	No	Yes	20 minutes	24 hours	Change every 14 days, sooner if cloudy.
Glutaraldehyde (Cidex®)	Varies (2–4%)	Add activator	Yes	Yes (vapors)	Yes	No	Yes	20 minutes at 25°C[d]	10 hours for Cidex®	Change every 14–28 days; sooner if cloudy.
Hydrogen Peroxide (30%)	6%	1 part 30% solution to 4 parts boiled water	Yes	Yes	No	Yes	No	20 minutes	Do not use	Change daily; sooner if cloudy.
CHEMICALS FOR DISINFECTION (alcohols and iodophors are not high-level disinfectants)										
Alcohol (ethyl or isopropyl)	60–90%	Use full strength	Yes (can dry skin)	Yes	No	No	No	Do not use	Do not use	If container (bottle) kept closed, use until empty.
Iodophors (10% povidone-iodine) (PVI)	Approximately 2.5%	1 part 10% PVI to 3 parts water	No	Yes	No	Yes	Yes	Do not use	Do not use	If container (bottle) kept closed, use until empty.

[a] All chemical disinfectants are heat- and light-sensitive and should be stored away from direct sunlight and in a cool place (<40°C).
[b] See **Tables 10-1 and 10-2** for instructions on preparing chlorine solutions.
[c] Only corrosive with prolonged (>20 minutes) contact at concentrations >0.5% if not rinsed immediately with boiled water.
[d] Different commercial preparations of Cidex and other glutaraldehydes are effective at lower temperatures (20°C) and for longer activated shelf life. **Always** check manufacturers' instructions.

Adapted from: Rutala 1996.

Products That Should Not Be Used as Disinfectants

Many antiseptic solutions are used incorrectly as disinfectants. Although antiseptics (sometimes called "skin disinfectants") are adequate for cleansing skin before surgical procedures, they are not appropriate for disinfecting surgical instruments and gloves. **They do not reliably destroy bacteria, viruses or endospores.** For example, Savlon (chlorhexidine gluconate with or without cetrimide), which is readily available worldwide, is often mistakenly used as a disinfectant.

Antiseptics that should not be used as disinfectants are:

- Acridine derivatives (e.g., gentian or crystal violet)
- Cetrimide (e.g., Cetavlon®)
- Chlorhexidine gluconate and cetrimide in various concentrations (e.g., Savlon)
- Chlorhexidine gluconate (e.g., Hibiscrub®, Hibitane®)
- Chlorinated lime and boric acid (e.g., Eusol®)
- Chloroxylenol in alcohol (e.g., Dettol®)
- Hexachlorophene (e.g., pHisoHex®)
- Mercury compounds

Mercury solutions (such as mercury laurel), although low-level disinfectants, **cause birth defects** and are too toxic to use as either disinfectants or antiseptics (Block 1991). (See **Appendix B** for details.)

Other products frequently used to disinfect equipment are 1–2% phenol (e.g., Phenol®), 5% carbolic acid (Lysol®) and benzalkonium chloride, a quaternary ammonium compound (Zephiran®). These are low-level disinfectants and should only be used to decontaminate environmental surfaces (e.g., floors or walls).

REFERENCES

Block SS (ed). 1991. *Disinfection, Sterilization and Preservation*, 4th ed. Lea & Febiger: Philadelphia.

Centers for Disease Control (CDC). 1987. Recommendations for prevention of HIV transmission in health care settings. *MMWR* 36(Suppl 2): 1S–18S.

Favero MS. 1985. Sterilization, disinfection, and antisepsis in the hospital, in *Manual of Clinical Microbiology*, 4th ed. Lennette EH et al (eds). American Society for Microbiology: Washington, DC, pp 129–137.

IPAS 1993. *Boiling IPAS Cannulas to Achieve High-Level Disinfection*. IPAS: Carrboro, NC, Scientific Report Summary.

Kobayashi H et al. 1984. Susceptibility of hepatitis B virus to disinfectants or heat. *J Clin Microbiol* 20(2): 214–216.

McIntosh N et al. 1994. *Practical Methods for High-Level Disinfection of Surgical Gloves*. Paper presented at American Public Health Association Annual Meeting. Session no. 2285, Washington, D.C., 31 October–4 November.

Perkins JJ. 1983. *Principles and Methods of Sterilization in Health Sciences*, 2nd ed. Charles C. Thomas Publisher Ltd.: Springfield, IL.

Russell AD, WB Hugo and GA Ayliffe. 1982. *Principles and Practice of Disinfection, Preservation and Sterilization*. Blackwell Scientific Publications: Oxford, England.

Rutala WA et al. 1998. Stability and bactericidal activity of chlorine solutions. *Infect Control Hosp Epidemiol* 19(5): 323–327.

Rutala WA. 1996. APIC guidelines for selection and use of disinfectants. *Am J Infect Control*, 24(4): 313–342.

Rutala WA. 1993. Disinfection, sterilization and waste disposal, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RP (ed). Williams & Wilkins: Baltimore, MD, pp 460–495.

Salle AJ. 1973. *Fundamental Principles of Bacteriology,* 7th ed. McGraw-Hill Book Company: New York.

Spaulding EH. 1939. Studies on chemical sterilization of surgical instruments. *Surg Gyne Obstet* 69: 738–744.

Tietjen LG, W Cronin and N McIntosh. 1992. High-level disinfection, in *Infection Prevention Guidelines for Family Planning Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 74–84.

Tietjen LG and N McIntosh. 1989. Infection prevention in family planning facilities. *Outlook* 7: 2–8.

World Health Organization (WHO). 1989. *Guidelines on Sterilization and High-Level Disinfection Methods Effective Against Human Immunodeficiency Virus (HIV)*. WHO: Geneva. AIDS Series 2.

Williams OB and CH Zimmerman. 1951. Studies on heat resistance III. *J Bacteriol* 61: 63.

THIRTEEN

PROCESSING LINEN

KEY CONCEPTS you will learn in this chapter include:

- Why careful handling and processing of soiled linen are important
- Which personal protective equipment to use and why
- How soiled linen should be collected and transported
- How soiled linen should be sorted, washed and dried
- How clean linen should be stored, transported and distributed

BACKGROUND

Although soiled linen may contain large numbers of microorganisms, there is little risk to health workers during linen processing. When worker-related infection has occurred, it often is the result of workers' not using gloves or washing their hands during or after collecting, transporting and sorting soiled items. To reduce the risk of contamination, staff at each healthcare facility should determine the best way to handle, process and store linens.

As the types and volume of services that hospitals and primary health clinics have expanded, so too has the need for clean linen on the wards and in housekeeping. In addition, surgical units, specialty areas (e.g., neonatal ICUs) and other departments such as anesthesiology, radiology and cardiology, where a variety of invasive medical procedures now are performed, have increased needs for linen items (caps, masks and gowns). As a consequence, in many large hospitals the laundering of linen increasingly is contracted out to companies specializing in this work. Regardless of where the soiled linen is processed, however, the infection prevention practices that are recommended to safely process linen are the same.

Remember: No additional precautions (e.g., pre-rinsing, labeling, separating or double bagging) are necessary, regardless of the patient's diagnosis, if Standard Precautions are used in all situations (Lynch et al 1997).

In smaller hospitals and clinics, however, housekeeping and cleaning staff will continue to be responsible for handling and processing soiled linen and other items. To do this job well, staff performing these tasks should be appropriately trained and regularly supervised. Without this, accidents will happen and staff will be at increased risk of exposure to infectious materials and acquiring work-related infections (Economics Report 1994).

DEFINITIONS

- **Detergent**. Cleaning agent that makes no antimicrobial claims on the label. Detergents (liquid or powder) are composed of a hydrophilic (water-seeking) component and a lipophilic (fat-seeking) component and can be divided into four types: anionic, cationic, amphoteric, and nonionic detergents.
- **Linens**. Cloth items used in healthcare facilities by housekeeping staff (bedding and towels), cleaning staff (cleaning cloths, gowns and caps) and surgical personnel (caps, masks, scrub suits, surgical gowns, drapes and wrappers) as well as by staff on specialty units such as ICUs and other units performing invasive medical procedures (e.g., anesthesiology, radiology or cardiology).
- **Occupational injury** or **infection**. Injury or infection acquired by healthcare staff while performing their normal duties.
- **Soaps** and **detergents (terms used interchangeably)**. Cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands. **Plain** soaps require friction (scrubbing) to mechanically remove microorganisms, while **antiseptic** (antimicrobial) soaps also kill or inhibit growth of most microorganisms.
- **Soiled** or **contaminated linen**. Linen from multiple sources within the hospital or clinic that has been collected and brought to the laundry for processing. All items, regardless of whether or not they are visibly dirty or have been used in a surgical procedure, must be washed and dried. For example, even though the sterile towel drapes contained in a surgical pack have not been used, they must be laundered before they can be sterilized (see **Chapter 5**).
- **Sorting**. Process of inspecting and removing foreign, and in some cases dangerous, objects (e.g., sharps or broken glass), from soiled linen before washing. This step is extremely important because soiled linen from the operating room or clinic occasionally contains sharps (e.g., scalpels, sharp-tipped scissors, hypodermic and suture needles and towel clips).

PROCESSING LINEN

Note: If utility gloves are not available, putting on two pairs of examination or reprocessed surgical gloves (double gloving) provides some protection for staff responsible for collecting, transporting and sorting soiled linen and other items.

Processing linen consists of all the steps required to collect, transport and sort soiled linen as well as to launder (wash, dry and fold or pack), store and distribute it. Safely processing linen from multiple sources is a complex process. The principles and key steps are listed in **Table 13-1**. Staff assigned to **collecting, transporting** and **sorting** soiled linen need to be especially careful. They should wear thick utility or heavy-duty household gloves to minimize the risk of accidental injury from a needlestick or other sharp object, including broken glass (see **Chapter 4**).

Staff responsible for washing soiled items should wear utility gloves, protective eyewear and plastic or rubber aprons.

Table 13-1. Principles and Key Steps in Processing Linen

- Housekeeping and laundry personnel should wear gloves and other personal protective equipment as indicated when collecting, handling, transporting, sorting and washing soiled linen.
- When collecting and transporting soiled linen, handle it as little as possible and with minimum contact to avoid accidental injury and spreading of microorganisms.
- Consider all cloth items (e.g., surgical drapes, gowns, wrappers) used during a procedure as infectious. Even if there is no visible contamination, the item must be laundered.
- Carry soiled linen in covered containers or plastic bags to prevent spills and splashes, and confine the soiled linen to designated areas (interim storage area) until transported to the laundry.
- Carefully sort all linen in the laundry area before washing. **Do not presort or wash linen at the point of use**.

USE OF PERSONAL PROTECTIVE EQUIPMENT

Listed in **Table 13-2** is the recommended personal protective equipment (PPE) for use by staff performing the various tasks associated with processing linens.

Table 13-2. Recommended Personal Protective Equipment for Processing Linen

TYPE OF PPE	WHEN TO WEAR
Gloves (preferably household utility gloves) and closed shoes that protect feet from dropped items (sharps) and spilled blood and body fluids	• Handling disinfectant solutions • Collecting and handling soiled linen • Transporting soiled linen • Sorting soiled linen • Hand washing soiled linen • Loading automatic washers
Plastic or rubber apron and protective eyewear	• Sorting soiled linen • Hand washing soiled linen • Loading automatic washers

COLLECTING, TRANSPORTING AND SORTING SOILED LINEN

Collecting and Transporting

After invasive medical or surgical procedures or when changing linen in patient rooms:

- Collect used linen in cloth or plastic bags or containers with lids. If linen is heavily contaminated with blood or body fluids, carefully roll the contaminated area into the center of the linen and place in a leakproof bag or container with a lid.
- Cloth bags are adequate for the majority of the patient care linen. They require the same processing as their contents.

- Handle soiled linen as little as possible and do not shake it. This helps prevent spreading microorganisms to the environment, personnel and other patients.
- It is not necessary to double-bag or use additional precautions for used linen from patients in isolation.
- Do not sort and wash soiled linens in patient care areas (CDC 1988; OSHA 1991).
- Collect and remove soiled linen after each procedure, and daily or as needed from patient rooms.
- Transport collected soiled linen in closed leakproof bags, containers with lids or covered carts to the processing area daily or more often as needed.
- Transport soiled linen and clean linen separately. If there are separate carts or containers available for soiled and clean linen, they should be labeled accordingly. If not, thoroughly clean the containers or carts used to transport soiled linen before using them to transport clean linen.

Note: Several studies have shown that there is no difference in the level of linen contamination from isolated and nonisolated patients (Maki, Alvarado and Hassemer 1986; Pugliese 1989; Weinstein et al 1989).

Sorting Soiled Linen

The processing area for soiled linen must be separate from other areas such as those used for folding and storing clean linen, patient care areas and food preparation areas. In addition there should be adequate ventilation and physical barriers (walls) between the clean and soiled linen areas.

Safe sorting of linen is extremely important. Sorting must be carefully performed because soiled linen (large drapes and towel drapes) from the operating room or other procedure areas not infrequently contain sharps (e.g., scalpels, sharp-tipped scissors, hypodermic and suture needles and sharp-tipped towel clips). In addition, bedding from patients' rooms may contain soiled dressings and be blood-stained or wet with other body fluids. These items must be handled carefully while wearing protective gloves, protective eyewear and plastic or rubber apron, and should be disposed of properly. Though infrequent, infections related to sorting have been attributed to failure of handwashing and proper use of PPE (McDonald 2002).

Remember: Disposable sharps (suture needles, scalpel blades and hypodermic needles) must be placed in sharps containers located near the point of use.

Soiled linen may also contain noninfectious items such as dentures, eyeglasses and hearing aids. These items pose no threat of infection and require no special handling.

LAUNDERING LINEN

Washing and Drying

All linen items (e.g., bed sheets, surgical drapes, masks, gowns) used in the direct care of a patient must be thoroughly washed before reuse. Decontamination prior to washing is **not necessary**, unless linen is heavily soiled and will be hand washed (repeated soaking of linen in chlorine,

even dilute solutions, will cause the fabric to deteriorate more quickly). Staff responsible for hand washing linen should use PPE as described in **Table 13-2**. In addition, workers should not carry wet, soiled linen close to their bodies even if they are wearing a plastic or rubber apron.

Remember: The storage time for soiled linen before washing is related to practical issues, such as available storage space and aesthetics, **not** to infection prevention concerns.

Hand Washing

STEP 1: Wash heavily soiled linen separately from nonsoiled linen.

STEP 2: Wash the entire item in water with liquid soap to remove all soilage, even if not visible:

Remember: Presoak in soap, water and bleach, **only** if linen is heavily soiled.

- Use warm water if available.
- Add bleach (e.g., 30–60 mL, about 2–3 tablespoons, of a 5% chlorine solution) to aid cleaning and bactericidal action.
- Add sour (a mild acid agent) to prevent yellowing of linen, if desirable.

STEP 3: Check the item for cleanliness. Rewash if it is dirty or stained.

STEP 4: Rinse the item with clean water.

Machine Washing

STEP 1: Wash heavily soiled linen separately from nonsoiled linen.

STEP 2: Adjust the temperature and time cycle of the machine according to manufacturer's instructions and the type of soap or other washing product being used. Both cold and hot water washing cycles that include bleach reduce bacterial counts in the linen.

Hot-water washing:[1]

- Use hot water above 71ºC (160°F) and soap to aid in loosening soil.
- Add bleach and sour as above.
- Adjust the time cycle of the machine according to the manufacturer's instructions.

Note: Uniforms and scrubsuits or gowns worn by housekeeping or cleaning staff can be safely laundered at home (i.e., home laundering does not increase the risk of infection to patients or staff) (Manangan 2001).

STEP 3: When the wash cycle is complete, check the linen for cleanliness. Rewash if it is dirty or stained. (Heavily soiled linen may require two wash cycles.)

[1] Lower temperatures or cold water washing are satisfactory if the cleaning products (type of soap or detergent, amount of bleach and other additives) are appropriate and used in proper concentrations. Using cold water also saves energy.

Drying, Checking and Folding Linen

For both hand and machine washed linens, the steps are the same.

STEP 1: Completely air or machine dry before further processing. Air dry in direct sunlight, if possible, keeping the fabric off the ground, away from dust and moisture.

STEP 2: After linen items are totally dry, check for holes and threadbare areas. If these are present, the item must be discarded or repaired before reuse or storage. (If there are any holes or many repaired areas, the item should not be used as a drape. It can be cut into pieces to be used as cleaning rags.)

Setting standards helps determine when drapes (lap sheets) or linen wrappers should be made into rags. For example, a drape should have no more than 5 patches per 1-foot (12 inches) square area or 20% of the drape covered with patches. Patches should be avoided because they increase the thickness of the linen item and decrease steam penetrability if sterilization is required.

Note: If surgical drapes are to be sterilized, do not iron. Ironing dries out the material, making auto-claving more difficult.

STEP 3: Clean and dry linen should be ironed as needed and folded. For example, if a **clean, dry drape** is acceptable, the drape can be ironed before placing it on a shelf or in a container for storage.

If **sterile linens** are required, prepare and sterilize wrapped packs as discussed in **Chapter 11** and **Appendix G**. The recommended guidelines for processing soiled linens are summarized in **Table 13-3**.

STORING, TRANSPORTING AND DISTRIBUTING CLEAN LINEN

Storing Clean Linen

- Keep clean linen in clean, closed storage areas.
- Use physical barriers to separate folding and storage rooms from soiled areas.
- Keep shelves clean.
- Handle stored linen as little as possible.

Transporting Clean Linen

- Clean and soiled linen should be transported separately.
- Containers or carts used to transport soiled linen should be thoroughly cleaned before used to transport clean linen. If different containers or carts are used to transport clean and soiled linen, they should be labeled.
- Clean linen must be wrapped or covered during transport to avoid contamination.

Distributing Clean Linen

- Protect clean linen until it is distributed for use.
- Do not leave extra linen in patients' rooms.
- Handle clean linen as little as possible.
- Avoid shaking clean linen. It releases dust and lint into the room.
- Clean soiled mattresses before putting clean linen on them.

Table 13-3. Guidelines for Processing Linens and Personal Protective Equipment (PPE)

ITEM	DECONTAMINATION	CLEANING	HIGH-LEVEL DISINFECTION	STERILIZATION
Protective eyewear (plastic goggles and face shields)	Wipe with 0.5% chlorine solution. Rinse with clean water. After each procedure or when is visibly soiled.	Wash with liquid soap and water. Rinse with clean water, then air or towel dry.[2] After each procedure or when visibly soiled.	Not necessary	Not necessary
Linens (caps, masks, scrubsuits or covergowns)	Not necessary. (Laundry staff should wear plastic aprons, gloves, and protective foot and eyewear when handling soiled items.)	Wash with liquid soap and water, removing all dirt particles. Rinse with clean water, air or machine dry.[2] Air-dried attire can be ironed before use.	Not necessary	Not necessary
Aprons (heavy plastic or rubber)	Wipe with 0.5% chlorine solution. Rinse with clean water. Between each procedure or each time they are taken off.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.[2]	Not necessary	Not necessary
Footwear (rubber shoes or boots)	Wipe with 0.5% chlorine solution. Rinse with clean water. At the end of the day or when visibly soiled.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.[2]	Not necessary	Not necessary
Surgical gowns, linen drapes and wrappers	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear when handling soiled items.)	Wash with liquid soap and water, removing all particles. Rinse with clean water, air or machine dry.[2]	Not practical	Preferred
Paper or disposable plastic items	Place in plastic bag or leakproof, covered waste container for disposal.			

[2] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

REFERENCES

Centers for Disease Control (CDC). 1988. Update: Universal precautions for prevention of transmission of HIV, HBV, and other bloodborne pathogens in health setting. *MMWR*: 37(24): 377.

Economics Report. 1994. In-house laundry/linen reprocessing: Who does it? *Health Facilities Management* 7(6): 126.

Lynch P et al. 1997. Barrier precautions: Reducing risks for transmission of organisms, in *Infection Prevention with Limited Resources*. ETNA Communications: Chicago, pp 87–92.

Maki DG, C Alvarado and C Hassemer. 1986. Double-bagging of items from isolation rooms is unnecessary as an infection control measure: A comparative study of surface contamination with single- and double-bagging. *Infec Control* 7(11): 535–537.

Manangan LP. 2001. Infection control dogma: Top 10 suspects. *Infect Control Hosp Epidemiol* 22(4): 243–247.

McDonald LL. 2002. Linen services, in *APIC Text of Infection Control and Epidemiology*. APIC: Washington, DC, pp 75-1–75-4.

Pugliese G. 1989. Isolating and double-bagging laundry: Is it really necessary? *Health Facility Management* 2(2): 16–20.

Occupational Safety and Health Administration (OSHA), US Department of Labor. 1991. Occupational exposure to bloodborne pathogens: Final rule. *Fed Regist* 56(235): 64004–64182.

Weinstein SA et al. 1989. Bacterial surface contamination of patient's linen: Isolation precautions versus standard care. *Am J Infect Control* 17(5): 264–267.

FOURTEEN

REPROCESSING DISPOSABLE (SINGLE-USE) ITEMS

KEY CONCEPTS you will learn in this chapter include:

- What the basis for reprocessing disposable (single-use) items is
- What the concerns regarding reprocessing disposable items are
- What appropriate guidelines for reprocessing disposable surgical gloves are
- What appropriate guidelines for recycling or reprocessing disposable syringes are

BACKGROUND

In the past, medical devices used in healthcare facilities were divided into two categories:

1. reusable devices or items intended to be cleaned, inspected and either high-level disinfected or sterilized for multiple reuses; and
2. disposable devices intended to be used once and then discarded.

The plastics revolution, however, has permanently changed the way healthcare is delivered by providing less expensive, disposable products for a multitude of purposes and procedures. Although disposables, such as single-use needles and syringes, minimize the risk of cross-contamination, plastics inadvertently also have increased healthcare costs for waste removal and environmental pollution resulting from incineration (burning).

Today more than 20 different types of plastics are used to produce hundreds of disposable medical devices, ranging in cost from a few cents (syringes) to more than a thousand dollars (electrophysiology catheters inserted into the heart to measure and correct rhythm disorders). Although designed for single use (disposable), approximately 30% of US hospitals currently reuse these catheters (Weinberg 1999). Thus, the issue really is not whether or not to reprocess, but what constitutes safe and appropriate reuse of disposable items.

How Safe Is Reprocessing

As a general principle, before starting a reuse program it should be demonstrated that the reprocessing of used, or previously opened but unused, items is as safe as reprocessing medical devices that are intended to be reusable (CHA 1996). For simple items such as disposable bedpans, dishware, Ambu bags, syringes and surgical gloves, reprocessing procedures are no different for the disposable item than for the reusable

equivalent (e.g., reusable versus disposable surgical gloves). Therefore, if there are appropriate guidelines and procedures for safely processing the reusable device or item, reprocessing the disposable will not be less safe and should result in the same quality product from a reprocessing perspective. Clearly, however, a reprocessed surgical glove, whether labeled reusable or disposable, may not be as safe in terms of protection (inapparent holes or tears) to the wearer, so guidelines for testing and the proper use of the reprocessed gloves must be in place if gloves are to be reused (see **Chapters 4** and **7**).

Benefits of Reprocessing

The most obvious benefit of reprocessing is the potential for cost savings, but for developing countries there is the added benefit of having a more dependable supply of items (e.g., surgical gloves and syringes) that only need to be replaced periodically. Other benefits to hospitals and the community are a reduction in the volume of medical waste, especially infectious waste, which is the most difficult and expensive to dispose of. For example, in some countries, the cost of properly transporting and disposing of waste is so high that nothing is done. As a consequence, all types of waste are dumped behind hospitals or clinics or partially burned in open pits. Finally, when a hospital or clinic reuses disposable devices, there is a saving to the environment in terms of pollution reduction, less incineration and less use of landfills and dump sites. Currently, American healthcare facilities send four billion pounds of waste to landfills and commercial incinerators each year (Dunn 2001)!

DEFINITIONS

- **Recycling**. Physical and/or chemical process that recovers the basic material in a product (e.g., paper from newspapers, aluminum from soft drink cans or plastic from disposable syringes) for reuse as a new or different product.
- **Reprocessing**. Decontaminating, disassembling (if necessary), cleaning, inspecting, testing, packaging, relabeling, and sterilizing or high-level disinfecting single-use devices (SUDs) after they have been used on a patient for their intended purpose. Reprocessing also is performed on SUDs that were removed from the package (or container) but not used on a patient or whose expiration date has passed.

REPROCESSING DISPOSABLE (SINGLE-USE) ITEMS

The use of disposable (single-use) items has increased steadily in recent years in nearly all countries, but especially in the US and Europe. **Table 14-1** shows the amount of waste produced by disposable items commonly used in a medium-sized hospital in Germany (Daschner 1993). Most of these disposables create additional environmental pollution and from an infection prevention perspective are unnecessary. For example, getting an infection from dishes used by a patient, even those with an acute respiratory or gastrointestinal infection, is highly unlikely.

Table 14-1. Waste Produced by Disposables per Year

DISPOSABLE	NUMBERS	WASTE (tons)	WASTE (%)
Kidney-shaped bowls	251,000	5.6	8
Gloves	2,100,000	19.4	28
Surgical drapes	54,000	6.7	9
Cleaning towels	194,000	7.0	10
Syringes	2,600,000	17.8	25
Dishes	388,000	6.1	9
Other items		7.9	11
Total		70.5	100%

Source: Dashner 1993.

As shown in **Table 14-2**, many reusable items (e.g., metal kidney basins) can safely replace disposables and are now beginning to be used even in the US. Fiscal constraints, budget cutbacks, managed care and data supporting the safety of reusing items are the driving forces behind the movement to replace or reuse disposables. In fact, in the US it is estimated that reprocessing could save $700 million per year if healthcare facilities took full advantage of the practice of reuse (Hawkins 1999; Selvey 2001).

Table 14-2. Disposables and Their Alternatives

ITEM	ALTERNATIVE
Single-use disposable Ambu bags	Reusable Ambu bags can be used for up to 8 years (they cost more up front, have reprocessing cost, but do not become waste for a long time)
Single-use disposable ventilator circuits	Reusable ventilator circuits
Single-use disposable gowns	Reusable cloth gowns
Single-use dishware	Reusable dishware, both crockery and cutlery
Disposable diapers (for young and old)	Reusable diapers
Single-use disposable pillows	Reusable pillows
Single-use disposable bedpans	Reusable plastic or steel bedpans
Single-use urinals	Reusable plastic or steel urinals
Single-use emesis basins	Reusable plastic or steel emesis basins
Single-use wash basins	Reusable plastic or steel wash basins
Single-use bowls	Reusable plastic or steel bowls
Disposable wash cloths	Reusable wash cloths
Disposable pitchers and cups	Reusable pitchers and cups

The situation in many developing countries, however, is quite different. Most disposable medical items have never been available because they are expensive and difficult to dispose of safely, especially plastic items. The two exceptions to this are disposable surgical gloves and disposable (plastic) syringes, which have rapidly replaced reusable products. As illustrated in **Table 14-1**, these two items alone create most of a moderate-sized hospital's medical waste—over 50% in fact!

Unlike the US, Canada and Western Europe, reprocessing of latex rubber surgical gloves is a standard practice in many countries because:

- supplies of new disposable gloves often are inadequate and stock outs not infrequent;
- reprocessing is not difficult and is inexpensive because low-cost labor is widely available; and
- reprocessed surgical gloves can be used not only in the operating room, but also as examination gloves, which generally are in short supply, for semi- and noncritical patient care activities.

REPROCESSING DISPOSABLE SURGICAL GLOVES

The risk in reprocessing is that reused surgical gloves contain more inapparent holes and tears than new ones. As a consequence, the wearer has less protection. But, as discussed in **Chapter 7**, even surgeons wearing new surgical gloves had a 14% blood-hand contact (Tokars et al 1995). Moreover in the US, the regulatory standard "acceptable" leak rate for the best quality latex rubber surgical or examination gloves is up to 4%! Wearing new gloves, therefore, does not guarantee that hands will be kept free of contaminating blood or body fluids, even in the absence of accidental breaks or tears. Moreover, as proposed in **Chapter 7**, double gloving with new gloves now is considered appropriate, given the current risk of exposure to HIV and HCV in many countries. Thus, sterilization (autoclaving) or steaming (high-level disinfection) of previously decontaminated and thoroughly cleaned surgical gloves can produce an acceptable product and, when combined with double gloving, constitutes an appropriate and cost-effective reuse of a disposable item.

In **Appendix C**, guidelines and detailed instructions are provided for the safe reprocessing of surgical gloves.

RECYCLING OR REPROCESSING DISPOSABLE (PLASTIC) SYRINGES AND HYPODERMIC NEEDLES

In countries with limited resources, the practice of collecting, selling and reusing syringes and hypodermic needles is a long-standing, logical response to scarcity and economics. Unfortunately, the individuals who scavenge landfills and public dumps for them, as well as the patients who

buy the cheaper "reprocessed" product, are at increased risk of infection with bloodborne pathogens. To date, attempts to discourage this practice have been universally unsuccessful.

Most developing country governments are overburdened and underfunded. As such, solutions to this problem that require functioning national systems for the management of medical waste and/or increased spending are unlikely to be implemented in the near future (Mujeeb et al 2003).[1] Thus, alternatives that recognize the economic value of used syringes are more likely to be successful in addressing this serious problem. Potential options include discarding the needles after decontamination and then either **recycling** the plastic syringe or **reprocessing** it according to recommended infection prevention practices.

Recycling Disposable Syringes

Recycling is a new, potential alternative for the safe disposal of plastic syringes that is appropriate for use in limited-resource settings. The other alternatives are incineration, encapsulation and safe burying (**Chapter 8**). In many countries, plastic recycling is a major industry. In developing countries, however, syringe recycling occurs primarily in the plasticware industry and is unregulated.

Of the alternatives, recycling is clearly more practical. Incineration produces potentially toxic emissions, including persistent organic pollutants in the case of low-temperature burning (see below) and is expensive. Encapsulation and safe burying not only are expensive, but also they do not reduce the ever-increasing volume of waste. And unlike recycling, neither one generates funds.

To make syringe recycling (and reprocessing as well) safer for scavengers and plastic workers, healthcare providers must consistently decontaminate assembled needles and syringes after use (**Appendix D**).[2] Implementing "point of use" decontamination also provides an added measure of protection to health workers, especially housekeeping and maintenance personnel, from acquiring a life-threatening disease (i.e., reduced risk that a needlestick will transmit HIV or other bloodborne viral infection). Furthermore, if the needles and syringes, as well as sharps containers, are not properly disposed of, the community is at **less risk** because the needles and syringes were decontaminated before being discarded.[3] Finally, the volume of waste would be significantly reduced, and the cost of disposal less, because the decontaminated needles and syringes could be treated as noninfectious waste. Thus, they could be sold for recycling (if available) or disposed of in dumps or public landfills.

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[1] While switching to autodisable syringes prevents reuse of the syringe, in many countries doing this will be expensive and logistically will take years to accomplish.

[2] Even the SoloShot FX™ autodisable syringe (**Chapter 7**) can be decontaminated because after use about 0.1 mL of chlorine solution can be drawn up. This volume is sufficient to completely fill the needle as well as cover the surface of the plunger and bottom of the syringe.

[3] HIV can survive in needles and syringes for more than 4 weeks at room temperature (Abdala et al 1999; Rich et al 1998).

Reprocessing Disposable Syringes (and Needles)

While r**eprocessing** disposable plastic syringes is a practical and economically viable alternative, to be safe it requires three conditions:

1. A sterile or high-level disinfected needle and syringe is used to give only a single injection.
2. After use, the assembled needle and syringe is decontaminated and placed in a sharps container.
3. The syringe, but preferably not the needle, is processed according to recommended infection prevention practices (thorough cleaning and either sterilization or high-level disinfection).

The rationale for reprocessing **only** the syringe, but **not** the needle, is the following:

- Contaminated needles are responsible for the injuries and the potential risk of acquiring a life-threatening disease.
- Needles are difficult to clean and sterilize or high-level disinfect, but syringes are not.
- Plastic syringes, many of which are made of polyvinyl chloride (PVC), contribute heavily to environmental pollution (i.e., converted to dioxins that are carcinogenic) when burned, even at high temperatures with scrubbers (NIHE 2002).

In **Appendix E**, guidelines and detailed instructions are provided for the safe:

- disposal of **both** needles and syringes,
- disposal of needles and processing of syringes, or
- processing of **both** needles and syringes in special situations.

Reprocessing Versus Disposal of Needles and Syringes

A major concern with reusing needles and syringes is the risk of transmitting HIV, HBV and HCV to patients if, after use, they are not reprocessed correctly, or several injections are given with the same needle and syringe (Drucker, Alcabes and Marx 2001; Simonsen et al 1999). To minimize this risk, in recent years disposable (single-use) plastic syringes and hypodermic needles, or one of the newer autodisable syringes that cannot be refilled, have been introduced in most countries. Clearly, wherever economically possible, disposable products should be used and safely disposed of after decontamination.[4]

Switching over to disposables, or the new autodisable syringes and needles, however, creates a new set of logistic and infection prevention

[4] While the autodisable syringes currently being used by USAID and UNICEF cannot be reused, their use does not address the risk to health workers, maintenance personnel and the community from accidental needlestick injuries unless they are decontaminated prior to disposal.

problems. For example, a clinic or hospital using only disposable or autodisable syringes must ensure that adequate supplies are available at all times. Without a continuous supply, services will be disrupted periodically. If a woman comes in for her 3-monthly injection of Depo Provera® (injectable contraceptive) only to find the clinic is out of syringes—this is a very real problem. An even more serious consequence would be if, rather than stopping services, the same disposable needle and syringe would then be used on more than one patient (i.e., the exact problem disposables were intended to solve) because procedures for safely reprocessing them are no longer in place.

A larger problem is **how to** safely dispose of used needles and syringes, both conventional and autodisposable, if resources for incineration, encapsulation or burying are not available. In many countries, used needles and syringes can be found lying discarded outside healthcare facilities and hospitals, or piled high in boxes in storage rooms. These used needles and syringes constitute an increasing health risk, especially to adults and children seeking items to sell, use or play with.

In summary, **reprocessing disposable syringes** constitutes an appropriate reuse of a disposable (single-use) device and significantly reduces infectious waste, as does reprocessing surgical gloves. Reprocessing syringes also limits costs because only new sterile needles need to be regularly resupplied. Moreover, reprocessing syringes further simplifies resupply problems because boxes of sterile needles, which are smaller and considerably less bulky, can be more easily and less expensively transported. Finally, reprocessing disposable syringes is income-generating, which for countries with limited resources is an important advantage.

Reprocessing used needles, however, represents an **inappropriate reuse of disposables** and is responsible for infections (Kane et al 1999; Phillips et al 1971; Simonsen et al 1999). In those situations where it is the only option available, it is critical that reprocessing be done as safely as possible using recommended infection prevention practices and processes (**Appendix E**).

REFERENCES

Abdala N et al. 1999. Survival of HIV-1 in syringes: Effects of temperature during storage. *J Acquir Immune Defic Syndr Hum Retrovirol* 20(1): 73–80.

Canadian Healthcare Association (CHA). 1996. *The Reuse of Single-Use Medical Devices: Guidelines for Healthcare Facilities.* Association for the Advancement of Medical Instrumentation: Arlington, VA.

Daschner F. 1993. The hospital and pollution: Role of the hospital epidemiologist in protecting the environment, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RP (ed). Williams & Wilkins: Baltimore, MD, pp 993–1000.

Drucker EM, PG Alcabes and PA Marx. 2001. The injection century: Consequences of massive unsterile injecting for the emergence of human pathogens. *Lancet* 358: 1989–1992.

Dunn D. 2001. Reprocessing single-use devices—the equipment connection. *AORN J* 75(6): 1143–1158.

Hawkins D. 1999. Risky recycling: That "disposable" catheter may have been used before. *US News World Report* 127(11): 62–64; 66–67.

Kane A et al. 1999. Transmission of hepatitis B, hepatitis C and human immunodeficiency viruses through unsafe injections in the developing world: Model-based regional estimates. *Bull World Health Organ* 77(10): 801–807.

Mujeeb SA et al. 2003. Recycling of injection equipment in Pakistan. *Infect Control Hosp Epidemiol* 24(2): 145–146.

Nightingale Institute for Health and the Environment (NIHE). 2002. *Nurses Can Make a Difference*. www.nihe.org.

Phillips I et al. 1971. *Pseudomonas cepacia (multicorans)* septicemia in an intensive care unit. *Lancet* 1(7695): 375–377.

Rich JD et al. 1998. Detection of HIV-1 nucleic acid and HIV-1 antibodies in needles and syringes used for non-intravenous injection. *AIDS* 12(17): 2345–2350.

Selvey D. 2001. Medical device reprocessing: Is it good for your organization? *Infec Control Today* 5: 22–26.

Simonsen L et al. 1999. Unsafe injections in the developing world and transmission of bloodborne pathogens: A review. *Bull World Health Organ* 77(10): 789–800.

Tokars JI et al. 1995. Skins and mucous membrane contacts with blood during surgical procedures: Risk and prevention. *Infect Control Hosp Epidemiol* 16(12): 703–711.

Weinberg N. 1999. Staying healthy blood money—hospitals are reusing devices they shouldn't. *Forbes* (22 March): 123–126.

FIFTEEN

TRAFFIC FLOW AND ACTIVITY PATTERNS[1]

KEY CONCEPTS you will learn in this chapter are:

- Why regulating traffic flow and defining activity patterns in hospitals and clinics are important
- How to design traffic flow and activity patterns in procedure, instrument processing and surgical areas
- What the traffic flow requirements are for these different areas

BACKGROUND

Microbial contamination is minimized by reducing the number of people permitted into an area and by defining the activities that take place there (Russell, Hugo and Ayliffe 1982).

Regulating the flow of visitors, patients and staff plays a central role in preventing disease transmission in healthcare facilities. Because the number of microorganisms in a designated area tends to be related to the number of people present and their activity, microbial contamination is expected—and found—to be high in areas such as waiting rooms and places where soiled surgical instruments and other equipment are initially processed.

An important objective of infection prevention is to minimize the level of microbial contamination in areas where patient care and instrument processing take place. Such areas include:

- **Procedure areas** where patients are examined and procedures (e.g., pelvic examinations, wound care management, blood drawing, immunizations, IUD insertions and removals, and normal childbirth) occur.
- **Surgical units** where major and minor operations are performed. The surgical unit also includes preoperative and recovery rooms as well as several other areas.
- **Work areas** where instruments are processed. These include dirty and clean areas where soiled instruments, equipment and other items are first cleaned and either high-level disinfected or sterilized and then stored.

[1] *Adapted from*: Tietjen, Cronin and McIntosh 1992.

It is important to direct activity patterns and traffic flow in these areas to keep contaminated areas separate from areas where procedures take place. Activities such as waste disposal, instrument processing and cleaning procedure areas should be carefully planned and organized to minimize the risk of infection to patients and healthcare workers. Equally important are designing and implementing traffic flow patterns that prevent soiled instruments and other items from crossing paths with cleaned, high-level disinfected or sterilized items.

Traffic flow also has to do with separating people who have, or are likely to have, communicable diseases from those who are at risk (susceptible). These people pose a great risk to susceptible patients and healthcare workers simply by being present in the same room; therefore, they need to be identified and quickly removed. For example, a child or teenager with a fever, an itchy rash on the head and body, and a negative history for chicken pox is best evaluated in the parking lot outside the hospital or clinic. Because triaging patients who may have a highly infectious disease involves staff quite different from those responsible for planning how to separate clean and dirty instruments, it is not addressed in this chapter. (Communicable disease triaging guidelines are fully described in **Chapter 21**.)

DEFINITIONS

- **At point of use**. Equipment, instruments and supply items are at the place where needed (e.g., sharps containers are placed within an arm's reach of where injections are being given).
- **Environmental controls**. Standards specifying procedures to be followed for the routine care, cleaning and disinfection of environmental surfaces, beds, bedrails, bedside equipment and other frequently touched surfaces.
- **Operating room**. Area or space where surgical procedures are performed.
- **Surgical unit**. Whole surgical area including the lockers and dressing rooms, preoperative and recovery rooms, peripheral support areas including storage space for sterile and high-level disinfected items and other consumable supplies, corridors leading to restricted areas, the operating room(s), scrub sink areas and the nursing station.

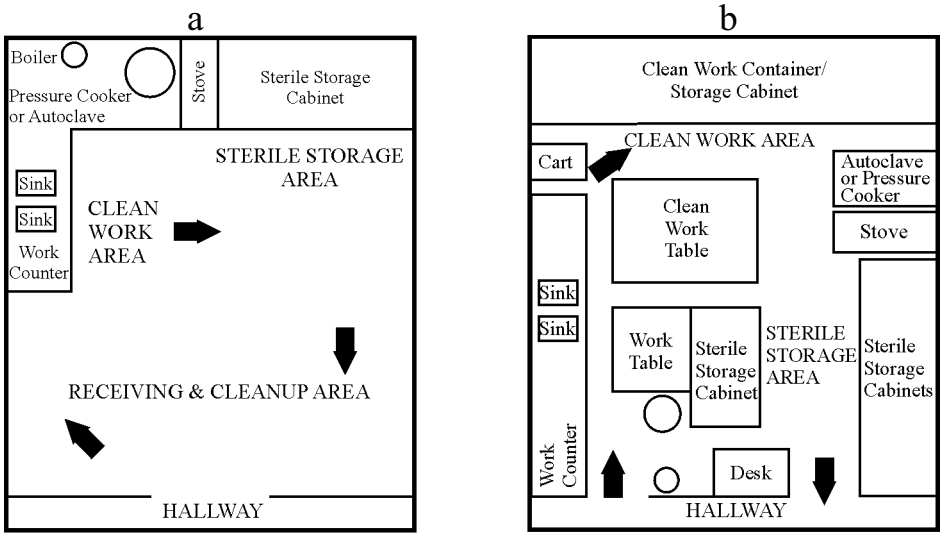
SPACE AND EQUIPMENT REQUIREMENTS

Healthcare facilities vary in the types of services they provide. For example, a rural clinic may offer only a few procedures (e.g., IUD insertion and removal, immunizations, antenatal care and minor surgery for suturing wounds or other trauma). Larger facilities (including district and referral hospitals) provide major and minor general surgical procedures in addition to ambulatory procedures. Regardless of the size of

the facility, however, the specific space and equipment requirements to perform a particular procedure generally do not vary.

In clinics where only minor procedures are performed, a **procedure room** with a handwashing sink is required for examining clients and performing procedures. A separate room with at least one sink for cleaning and an area for processing instruments and other items is also desirable (**Figure 15-1a**). Ideally, the processing area should include more than one room (e.g., a dirty room for receiving dirty instruments and a clean room for final processing and storage). If only a single room is available (**Figure 15-1a**), soiled equipment should be received and cleaned in an area of the room well away from where equipment is high-level disinfected or sterilized and stored.

Figure 15-1a and b. Floor Plans for Instrument Cleaning, High-Level Disinfecting and Sterilizing Areas in a Clinic and Larger Facility



Design for small clinic with minimal service space available

Design for larger facility or where volume is greater

Source: SEARO/WHO 1988.

Although the space requirements for performing various minor surgical procedures may not be different, depending on the classification of the procedure (semicritical or critical), the instrument processing requirements (high-level disinfection or sterilization) may be quite different. Inserting or removing an IUD, for example, is classified as a semicritical procedure involving intact mucous membranes, and the site (vagina and cervix) is not normally sterile, nor can it be made so (Spaulding 1968). By contrast, inserting a laparoscope into the abdomen is classified as a critical procedure because tissues that are normally sterile are being touched. For the former, either sterile or high-level disinfected instruments are acceptable, but for the latter the preferred final processing is sterilization (see

Chapters 1 and **9**).[2] Therefore, because of the need for sterile metal instruments with laparoscopy, an additional separate area for final processing (high-pressure sterilization by autoclaving) is desirable (**Figure 15-1b**). This is especially important if the volume of services is high (five or more procedures per day).

Note: Instruments should not be processed in the procedure room; nor should the handwashing sink be used for instrument cleaning.

The space, equipment and need for well-defined traffic flow and activity patterns become progressively more complex as the type of surgical procedure changes from general surgery and obstetrics to open heart surgery. As a guide, the space requirements for the types of surgery typically performed at district hospitals are roughly the same as for a busy surgicenter or polyclinic. These include:

- Changing room and scrub area for clinic staff
- Preoperative area where clients are examined and evaluated prior to surgery
- Operating room
- Recovery area for patient observation after surgery (may be combined with the preoperative area)
- Processing area for cleaning and sterilizing or high-level disinfecting instruments and other items
- Space for storing sterile packs and/or high-level disinfected containers of instruments and other items

MINIMIZING MICROBIAL CONTAMINATION

The recommended infection prevention practices for minimizing microbial contamination of specific areas in healthcare facilities are briefly described below.

Procedure Area

- Limit traffic to authorized staff and patients at all times.
- Permit **only** the patient and staff performing and assisting with procedures in the procedure room (family members should be limited with obstetrical procedures).
- Patients can wear their own clean clothing.
- Staff should wear attire and personal protective equipment (PPE) according to procedures performed.
- Have a covered container filled with a 0.5% chlorine solution for immediate decontamination of instruments and other items once they are no longer needed.

[2] Because laparoscopes are heat-sensitive, they can only be sterilized using chemical sterilants, such as formaldehyde or glutaraldehydes.

- Have a leakproof, covered waste container for disposal of contaminated waste items (cotton, gauze, dressings) at point of use.
- Have a puncture-resistant container for safe disposal of sharps (e.g., used suture needles, hypodermic needles and syringes, and disposable scalpel blades) at point of use.
- Have storage space in procedure rooms for clean, high-level disinfected and sterile supplies. (Storage shelves should be enclosed to minimize dust and debris collecting on stored items.)

Surgical Unit

The surgical unit is often divided into four designated areas, which are defined by the activities performed in each—unrestricted area, transition zone, semirestricted area and restricted area. Environmental controls and use of surgical attire increase as one moves from unrestricted to restricted areas. Moreover, staff with respiratory or skin infections and uncovered open sores should not be allowed in the surgical unit.

Note: Post signs in each area to clearly indicate the appropriate environmental control and surgical attire required.

Unrestricted Area

This area is the entrance from the main corridor and is isolated from other areas of the surgical unit. This is the point through which staff, patients and materials enter the surgical unit.

Transition Zone

This area consists primarily of dressing rooms and lockers. It is where staff put on surgical attire that allows them to move from unrestricted to semirestricted or restricted areas in the surgical unit. Only authorized staff should enter this area.

Semirestricted Area

This is the peripheral support area of the surgical unit and includes preoperative and recovery rooms, storage space for sterile and high-level disinfected items, and corridors leading to the restricted area. Support activities (e.g., instrument processing and storage) for the operating room occur here.

- Limit traffic to authorized staff and patients at all times.
- Have a work area for processing of clean instruments.
- Have storage space for clean and sterile or high-level disinfected supplies with enclosed shelves to minimize dust and debris collecting on stored items.
- Have doors limiting access to the restricted area of the surgical unit.
- Staff who work in this area should wear surgical attire and a cap.

Note: Flipflops or sandals should not be worn as they provide no protection from dropped sharps.

- Staff should wear clean, closed shoes that will protect their feet from fluids and dropped items.

Restricted Area

This area consists of the operating room(s) and scrub sink areas.

Note: Never store instruments and other items in the operating room.

- Limit traffic to authorized staff and patients at all times.
- Keep the door closed at all times, except during movement of staff, patients, supplies and equipment.
- Scrubbed staff must wear full surgical attire and cover head and facial hair with a cap and mask.
- Staff should wear clean, closed shoes that will protect their feet from fluids and dropped items.
- Masks are required when sterile supplies are open and scrubbed staff are operating.
- Patients entering the surgical unit should wear clean gowns or be covered with clean linen, and have their hair covered.
- Patients do not need to wear masks during transport (unless they require airborne precautions).

Operating Room(s)

- Enclose the operating room to minimize dust and eliminate flies; central air conditioning is preferred. (If windows are the only ventilation, provide tight-fitting screens.)
- The operating room should be located away from areas of the hospital or healthcare facility that are heavily traveled by staff and patients.

Before surgical procedures:

- Place a clean, covered container filled with a 0.5% chlorine solution or other locally available and approved disinfectant for immediate decontamination of instruments and other items once they are no longer needed.
- Place a plastic bag or leakproof, covered waste container for contaminated waste items (cotton gauze, old dressings).
- Place a puncture-resistant container for the safe disposal of sharps (e.g., suture needles, hypodermic needles and syringes, and disposable scalpel blades) at the point of use but without contaminating the sterile field.
- Place a leakproof, covered waste container for soiled linen away from sterile items.

- Organize tables, Mayo and ring stands side by side in an area away from the traffic patterns and at least 45 cm (18 inches) from walls, cabinets and other nonsterile surfaces.
- Place a clean sheet, a lift sheet and armboard covers on the operating room bed.
- Check and set up suction, oxygen and anesthesia equipment.
- Place supplies and packages that are ready to open on the tables, not on the floor.
- Mayo stand and other nonsterile surfaces that are to be used during the procedure should be covered with a sterile towel or cloth.

During surgical procedures:

- Limit the number of staff entering the operating room only to those necessary to perform the procedure and to patients (family members as needed). Make the surgical team self-sufficient so that outside help is not required.
- Keep the doors closed at all times, except during movement of staff, patients, supplies and equipment.
- Keep the number of people and their movement to a minimum; the numbers of microorganisms increase with activity.
- Keep talking to a minimum in the presence of a sterile field.
- Scrubbed staff should wear full surgical attire, including:

  - a clean scrub suit covering bare arms (one or two pieces); if a two-piece pantsuit is worn, the top of the scrub suit should be tucked into the pants;
  - a clean surgical cap that covers the head;
  - clean, closed shoes that protect the feet from fluids or dropped items; and
  - sterile (or high-level disinfected) surgical gloves, protective eyewear and a mask covering the mouth, nose and any facial hair.

Note: Healthcare personnel do not need to wear cover-gowns when leaving the operating room (Manangan et al 2001).

Note: If splashes or spills of blood or amniotic fluid are expected, wear a faceshield and plastic or rubber apron.

- Scrubbed staff should keep their arms and hands within the operative field at all times and touch only sterile items or areas.
- Nonscrubbed staff should wear surgical attire, including:

  - long sleeved jackets banded at the wrist and that are closed during use;
  - a clean surgical cap that covers the head;
  - clean, closed shoes that protect the feet from fluids or dropped items; and
  - a mask covering the mouth, nose and any facial hair.

- Nonscrubbed staff should stay at the periphery of the operating room, keeping their distance from sterile areas. They should not lean or reach over the operative field.
- Clean accidental spills or contaminated debris in areas outside the surgical field with a 0.5% chlorine solution as promptly as possible. (A nonscrubbed staff member wearing utility gloves should do this.)

After surgical procedures, nonscrubbed staff wearing utility gloves should:

- Collect all waste and remove it from the room in closed leakproof containers.
- Close and remove puncture-resistant containers when they are three quarters full.
- Remove covered containers with a 0.5% chlorine solution with instruments and surgical gloves from the room.
- Remove soiled linen in closed leakproof containers.
- Remove waste, soiled linen, soiled instruments and equipment, and supplies that have been opened but not used, in an enclosed cart or in a leakproof, covered waste container. (Be sure that these items do not re-enter the restricted area.)

Work Area

According to the size and type of the healthcare facility, the work area for processing instruments (e.g., the Central Supply Department or CSD) may be part of or connected to the surgical unit, or it may be an independent area somewhere away from the surgical unit.

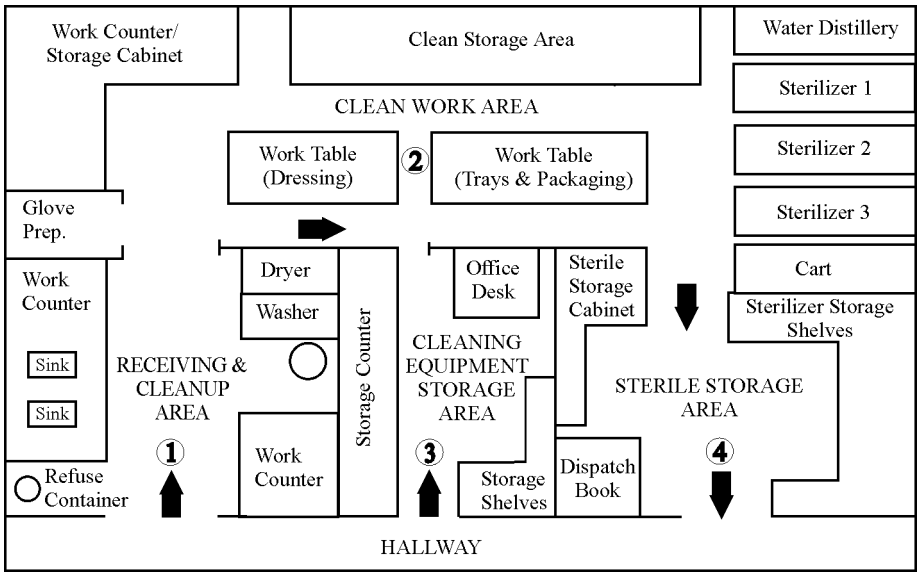
This is the area where instruments, surgical gloves and equipment are processed, and where staff should be specially trained in handling and processing and storing instruments, equipment and other clean, sterile or high-level disinfected items. The CSD is considered a semi-restricted area, so all the recommendations for traffic patterns and proper attire described above should be followed.

Remember: Permit only authorized personnel to enter this area.

A CSD consists of four areas, as shown in **Figure 15-2**. These areas are:

1. the “dirty” receiving/cleanup area,
2. the “clean” work area,
3. the cleaning equipment storage area, and
4. the sterile or high-level disinfected storage area.

Figure 15-2. Floor Plan for a Central Supply Department in a Hospital



Following surgery, decontaminate instruments, surgical gloves and other items by placing them in a plastic container filled with a 0.5% chlorine solution at the point of use. Cover the container and transport it to the CSD or designated instrument and equipment processing area. Alternatively, place soiled instruments in their original sterile wrap and transport them to the CSD where they can be immediately decontaminated before further processing.

Note: Develop flow patterns to help ensure that contaminated items never come in contact with clean, disinfected or sterile items.

Separate the "dirty" receiving/cleanup area (1) from the "clean" work area (2) with a physical barrier (wall and door). If this is not possible, use a screen or paint a red line on the floor to designate separation between areas.

The function and equipment requirements for the four areas of a typical CSD are summarized below.

"Dirty" Receiving/Cleanup Area (1)

Remember: Staff in the receiving/cleanup area should wear plastic aprons, utility gloves and safety goggles or face shields to protect themselves from spills and splashes.

In this area soiled items are received, disassembled and washed, rinsed and dried.

The "dirty" receiving/cleanup area should have:

- a receiving counter;[3]
- two sinks if possible (one for cleaning and one for rinsing) with a clean water supply; and
- a clean equipment counter for drying.

[3] If it is not possible to decontaminate instruments and other items in procedure or operating rooms, a decontamination counter is needed for this step.

"Clean" Work Area (2)

In the clean work area, cleaned items are:

- inspected for flaws or damage;
- packaged (if indicated), and either sterilized or high-level disinfected; and
- sent for storage as packaged or air dried and placed in a sterile or high-level disinfected container.

Note: Staff entering the clean work area should wear clean cover gowns.

The clean work area should have:

- a large work table;
- shelves for holding clean and packaged items; and
- a high-pressure steam sterilizer, a dry-heat oven, a steamer or a boiler.

Clean Equipment Storage Area (3)

Store clean equipment in this area. CSD staff also should enter the CSD through this area. Equip the area with:

- shelves (preferably enclosed) for storing clean equipment, and
- an office or desk for record keeping.

Sterile or High-Level Disinfected Storage Area (4)

Store sterilized packs and covered sterile or high-level disinfected containers in this area. This area should be separated from the central sterile supply area.

Note: Unwrapped objects must be used immediately.

- Limit access to the storage area and/or store items in closed cabinets or shelves. (Enclosed shelves or cabinets are preferred as they protect packs and containers from dust and debris. Open shelves are acceptable if the area has limited access, and housekeeping and ventilation practices are controlled.)
- Keep the storage area clean, dry, dust-free and lint-free by following a regular housekeeping schedule.
- Packs and containers with sterile or high-level disinfected items should be stored 20 to 25 cm (8 to 10 inches) off the floor, 45 to 50 cm (18 to 20 inches) from the ceiling and 15 to 20 cm (6 to 8 inches) from an outside wall.
- Do not use cardboard boxes for storage. (Cardboard boxes shed dust and debris and may harbor insects.)

- Date and rotate the supplies (first in, first out). This process serves as a reminder that the package is susceptible to contamination and conserves storage space, **but it does not guarantee sterility**.
- Packs will remain sterile as long as the integrity of the package is maintained.
- Sterile or high-level disinfected containers remain so until they are opened.
- Dispense sterile and high-level disinfected articles from this area.

Shelf Life (Belkin 1997a; Belkin 1997b)

- The shelf life of a packaged sterile item is event-related and not time-related. An event can compromise the integrity and effectiveness of the package.
- Events that can compromise or destroy package sterility include multiple handling, loss of package integrity, moisture penetration and airborne contamination.
- Sterility is lost when the package has tears in the wrapper, has become wet, has been dropped on the floor, has dust on it or is not sealed.
- The shelf life of a sterile package will depend on the quality of packing, conditions during storage and transport, and the amount of handling prior to use.
- Sealing sterile packs in plastic bags can help prevent damage and contamination.

Remember: Touch or handle sterile packages as little as possible.

- Most contaminating events are related to excessive or improper handling of the packages. The ideal number of times an item should be handled is three:

1. when removing it from the sterilizer cart and placing on a storage shelf,
2. when transporting it to the place where it is to be used, and
3. when selecting it to be opened for use.

Factors that can destroy sterility or compromise the efficiency of the packaging material to act as a bacterial barrier are:

- dust;
- moisture;
- holes, breaks, rupture of seals; and
- opening the package.

Before using any item that has been stored, check the package to be sure it is not dirty, wet or damaged.

Handling and Transporting Instruments and Other Items

- Keep clean and high-level disinfected or sterile instruments and other items separate from soiled equipment and waste items. Do not transport or store these items together.
- Transport high-level disinfected and sterile instruments and other items to the procedure or operating room in a closed cart or container with a **cover** to prevent contamination.
- Remove supplies from all shipping cartons and boxes before bringing such supplies into the procedure room, the operating room or the clean work area of the CSD. (Shipping boxes shed dust and harbor insects that may contaminate these areas.)
- Transport soiled supplies and instruments to the receiving/cleanup area of the CSD in leakproof, covered waste containers.
- Transport contaminated waste to the disposal site in leakproof, covered waste containers.

Note: If supplies are being delivered to the surgical area, one person standing outside should pass them through the door to a person inside the operating room.

(For additional information regarding handling and managing waste items, see **Chapter 8**.)

REFERENCES

Belkin NL. 1997a. *Textiles of Today: Their Influence on Sterilization and Shelf-Life, Part I.* www.cea.purdue.edu/IAHCSMM/25LESSON.HTM.

Belkin NL. 1997b. *Textiles of Today: Their Influence on Sterility and Shelf-Life, Part II*. www.cea.purdue.edu/IAHCSMM/26LESSON.HTM.

Manangan LP et al. 2001. Infection control dogma: Top 10 suspects. *Infect. Control Hosp Epidemiol* 22(4): 243–247.

Russell AD, WB Hugo and GA Ayliffe. 1982. *Principles and Practice of Disinfection, Preservation and Sterilization*. Blackwell Scientific Publications: Oxford, England.

South East Asia Regional Office (SEARO), World Health Organization (WHO) 1988. *A Manual on Infection Control in Health Facilities*. SEARO: New Delhi, India, pp 39–42.

Spaulding EH. 1968. Chemical disinfection of medical and surgical materials, in *Disinfection, Sterilization and Preservation*. Lawrence CA et al (eds). Lea & Febiger: Philadelphia.

Tietjen LG, W Cronin and N McIntosh. 1992. Traffic flow, in *Infection Prevention Guidelines for Family Planning Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 85–96.

SIXTEEN

HOUSEKEEPING

KEY CONCEPTS you will learn in this chapter include:

- Why housekeeping in hospitals and clinics is important
- What the general principles of cleaning are
- How to prepare disinfectant cleaning solutions
- When and how to clean low- and high-risk areas
- How to clean spills of blood or other body fluids
- How to clean housekeeping equipment

BACKGROUND

> **"Accumulation of dust, soil, and microbial contaminants on environmental surfaces is both aesthetically displeasing and a potential source of nosocomial infections. Effective and efficient cleaning methods and schedules are, therefore, necessary to maintain a clean and healthy environment in healthcare settings." (Chou 2002)**

Housekeeping refers to the general cleaning of hospitals and clinics, including the floors, walls, certain types of equipment, tables and other surfaces. The purpose of general housekeeping is to:

- reduce the number of microorganisms that may come in contact with patients, visitors, staff and the community; and
- provide a clean and pleasant atmosphere for patients and staff.

Most areas in hospitals and clinics are low-risk, such as waiting rooms and administrative offices, and can be cleaned using only soap and water. In high-risk areas where heavy contamination is expected, such as toilets and latrines, or for blood or body fluid spills, a disinfectant such as 0.5% chlorine or 1% phenol should be added to the cleaning solution (SEARO 1988). Using a disinfectant in addition to soap and water is also recommended in other high-risk areas such as operating rooms, pre- and postoperative recovery areas and intensive care units (ICUs).

In addition, patient rooms, especially those items that might be touched barehanded by patients and staff, should be cleaned using a disinfectant solution to minimize the risk of infection. For example, McFarland et al (1989) found that when patients who did not have *Clostridium difficile* were

admitted to a room previously occupied by a patient with *C. difficile*, the risk for the new patient increased several fold—even though staff were correctly using precautions to prevent cross-contamination.[1]

If the purpose of housekeeping as stated above is to be achieved, it is important that housekeeping staff be trained to perform their assigned tasks and are supervised on a regular basis. As part of their training, it is important that housekeeping staff:

- understand the risk of exposure to contaminated items and surfaces when performing environmental cleaning procedures; and
- follow recommended policies and guidelines, including the use of appropriate personal protective equipment (PPE).

The general principles for cleaning hospitals and clinics and other healthcare facilities are summarized in **Table 16-1**.

Table 16-1. General Principles of Cleaning

- **Scrubbing (frictional cleaning)** is the best way to physically remove dirt, debris and microorganisms.
- **Cleaning** is required **prior** to any disinfection process because dirt, debris and other materials can decrease the effectiveness of many chemical disinfectants.
- Cleaning products should be selected on the basis of their **use, efficacy, safety** and **cost**.
- **Cleaning** should always progress from **the least soiled areas to the most soiled areas** and from **high to low areas**, so that the dirtiest areas and debris that fall on the floor will be cleaned up last.
- **Dry sweeping, mopping and dusting** should be avoided to prevent dust, debris and microorganisms from getting into the air and landing on clean surfaces. Airborne fungal spores are especially important as they can cause fatal infections in immunosuppressed patients (Arnow et al 1991).
- **Mixing (dilution) instructions should be followed** when using disinfectants. (Too much or too little water may reduce the effectiveness of disinfectants.)
- Cleaning methods and written cleaning schedules should be based on the **type of surface**, **amount and type of soil present** and the **purpose of the area**.
- **Routine cleaning** is necessary to maintain a standard of cleanliness. Schedules and procedures should be consistent and posted.

DEFINITIONS

- **Cleaning solution**. Any combination of soap (or detergent) and water, with or without a chemical disinfectant, used to wash or wipe down environmental surfaces such as floors, chairs, bench tops, walls and ceilings.
- **Disinfectant**. Chemical that destroys or inactivates microorganisms. Disinfectants are classified as low-, intermediate- or high-level

[1] *C. difficile* is an excellent marker for organisms such as enterococci that persist in the environment.

depending on their ability to kill or immobilize some (low- or intermediate-level) or all (high-level) microorganisms (but not all spores). Phenols, chlorine or chlorine-containing compounds and QUATs are classes of disinfectants frequently used to clean noncritical surfaces such as floors, walls and furniture.

- **Disinfectant cleaning solution**. Products that are a combination of a detergent (soap) and a chemical disinfectant. Not all detergents and disinfectants are compatible. Several combinations are available commercially or can be prepared, such as alkaline detergents with chlorine compounds, alkaline detergents with quaternary ammonium compounds (QUATs) or other nonionic surfactants, and acid detergents with iodophors.
- **Environmental controls**. Standards specifying procedures to be followed for the routine care, cleaning and disinfection of environmental surfaces, beds, bedrails, bedside equipment and other frequently touched surfaces.
- **Environmental hygiene**. Process of maintaining a clean, healthy and pleasing patient and work environment.
- **Sanitizer**. Chemical that reduces the number of bacterial contaminants to safe levels on inanimate objects based on public health requirements (i.e., a chemical that kills 99.999% of the specific test bacteria in 30 seconds under the conditions of the test).
- **Soaps** and **detergents (terms used interchangeably)**. Cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands. **Plain** soaps require friction (scrubbing) to mechanically remove microorganisms; **antiseptic** (antimicrobial) soaps kill or inhibit the growth of most microorganisms.
- **Sterilants**. Chemicals used to destroy all forms of microorganisms, including endospores. Most sterilants are also high-level disinfectants when used for a shorter period of time. Sterilants are used only on inanimate objects (e.g., surgical instruments) that are used in semicritical and critical areas (e.g., surgery). Sterilants are not meant to be used for cleaning environmental surfaces.
- **Surfactant**. Agent that reduces the surface tension of water or the tension at the interface between water and another liquid; a wetting agent found in many sterilants and disinfectants.
- **Type of detergent**: Commercial cleaning product (liquid or powder) that are composed of a hydrophilic (water-seeking) component and a lipophilic (fat-seeking) component and can be divided into four types: anionic, cationic, amphoteric and nonionic detergents.

HOW TO SELECT A CLEANING PRODUCT

Different types of cleaning products are available—liquid soaps and detergents, disinfectants, combinations (detergent and disinfectant) and sanitizers—and each type has different properties. An ideal cleaning product should accomplish the following:

- Suspension of fats (suspend fats in water)
- Saponification of fats (make fats water-soluble)
- Surfaction (decrease surface tension of water and allow greater penetration of the agent into the dirt or soil)
- Dispersion (break up of soil into small particles)
- Protein destruction (break up proteins)
- Softening the water (removal of calcium and magnesium)

When selecting a disinfectant or other cleaning product, consider the following factors:

- Intended use
- Efficacy
- Acceptability
- Safety
- Cost

In settings where resources are limited, it is important not to waste money on expensive cleaning products that are unnecessary. Where the volume of intended use is high, preparing cleaning solutions from bulk products should be considered. For smaller facilities, it may be necessary to purchase commercial products for use in cleaning high-risk areas, such as operating rooms, to ensure that cleaning meets the requirements for the area. What is important is that the decision as to what product(s) to buy or use is not left to chance.

HOW TO PREPARE A DISINFECTANT CLEANING SOLUTION

A disinfectant cleaning solution is one that contains both a disinfectant and a detergent (soap).

Precautions When Using Chlorine Solutions

Although chlorine-containing solutions (sodium hypochlorite) are excellent, inexpensive disinfectants, they should **not** be mixed with cleaning solutions containing an acid (e.g., phosphoric acid), ammonia or ammonium chloride (NH_2Cl). Doing this will release chlorine gas and other by-products that can

result in temporary illness (nausea, tearing, headache or shortness of breath) to staff breathing fumes in a poorly ventilated area (CDC 1991).

To find out if a cleaning solution contains ammonia, first check the label. If it does not say there is ammonia, you may be able to detect ammonia when opening the product by its pungent, burning smell.

If you are exposed to chlorine gas or ammonium chloride or other unpleasant (noxious) gases with strong odors, leave the room or area immediately until the room can be completely ventilated.

Instructions

STEP 1: Prepare a 0.5% chlorine solution from liquid concentrates (see **Table 10-1** for directions) or from chlorine compounds (see **Table 10-2**). Alternative disinfectants that can be used include 1–2% phenols or 5% carbolic acid (e.g., Lysol®).

STEP 2: Add enough detergent to the 0.5% chlorine solution or other disinfectant to make a mild, soapy cleaning solution.

CLEANING METHODS

In general, written schedules and procedures for cleaning in each specific area should be available and posted. Cleaning should start with the least soiled area and move to the most soiled area and from high to low surfaces. Common methods of cleaning are briefly described below:

Note: **Do not use disinfectant fogging** (e.g., fumigation with dilute formaldehyde (formalin) solutions to reduce microbial contamination of environmental surfaces such as walls, ceilings and floors (CDC 1988). It is not effective, is time-consuming (requires 24 hours) and the fumes are toxic (irritating to mucous membranes of the nose and eyes). Scrubbing with a disinfectant and cleaning is a safer, quicker and more effective way to reduce microbial contamination on these surfaces.

Wet mopping is the most common and preferred method to clean floors.

- **Single-bucket (basin) technique**: One bucket of cleaning solution is used. The solution must be changed when dirty. (The killing power of the cleaning product decreases with the increased load of soil and organic material present.)
- **Double-bucket technique**: Two different buckets are used, one containing a cleaning solution and the other containing rinse water. The mop is always rinsed and wrung out before it is dipped into the cleaning solution. The double-bucket technique extends the life of the cleaning solution (fewer changes are required), saving both labor and material costs.
- **Triple-bucket technique**: The third bucket is used for wringing out the mop before rinsing, which extends the life of the rinse water.

Flooding followed by wet vacuuming is recommended in the surgical suite, if possible. This process eliminates mopping, thus minimizing the spread of microorganisms. This method increases the contact time of disinfectants with the surface to be cleaned, but it is necessary to leave the floor wet for several

minutes. (Flooding is best done at night or at times when foot traffic is minimal.)

Dusting is most commonly used for cleaning walls, ceilings, doors, windows, furniture and other environmental surfaces.

- Clean cloths or mops are wetted with cleaning solution contained in a basin or bucket. The double-bucket system minimizes the contamination of the cleaning solution.
- Dry dusting should be avoided and dust cloths and mops should never be shaken to avoid the spread of microorganisms.
- Dusting should be performed in a systematic way, using a starting point as a reference to ensure that all surfaces have been reached.
- When doing high dusting (ceiling tiles and walls), check for stains that may indicate possible leaks. (Leaks should be repaired as soon as possible because moist ceiling tiles provide a reservoir for fungal growth.)

Dry vacuuming is only recommended for cleaning of carpets.

USE OF PERSONAL PROTECTIVE EQUIPMENT

Table 16-2 lists the recommended PPE for use by housekeeping staff when performing the various tasks.

Table 16-2. Recommended Personal Protective Equipment for Housekeeping Tasks

TYPE OF PPE	WHEN USED
Gloves (preferably household utility gloves) Shoes that protect the feet from accidentally dropped items and blood and body fluids	• Handling disinfectant cleaning solutions • Cleaning patient care areas • Cleaning heavily contaminated areas • Handling soiled linen • Handling soiled items and instruments • Handling or disposing of waste
Plastic or rubber apron, mask and protective eyewear	• When spills or splashes are expected

SCHEDULE AND PROCEDURES FOR SPECIFIC AREAS

Note: Environmental surfaces are rarely associated with disease

Housekeeping schedules should be planned, **written** and closely followed. Cleaning schedules should be developed according to the needs of each area.

- **Walls, windows, ceilings and doors, including door handles**: Spot clean when visibly dirty with a damp cloth, detergent and water. In general, routine damp dusting is adequate for these areas (disinfection is unnecessary). These surfaces are rarely heavily contaminated with

microorganisms, as long as the surfaces remain dry and intact (Russell, Hugo and Ayliffe 1982).

- **Chairs, lamps, tables, tabletops, beds, handrails, grab bars, lights, tops of doors and counters**: Wipe daily and whenever visibly soiled with a damp cloth, containing disinfectant cleaning solution. A disinfectant should be used when contamination is present, such as for blood or other body fluid spills as described below.
- **Noncritical equipment** (e.g., stethoscopes and blood pressure cuffs): Wipe daily and whenever visibly soiled with a damp cloth, detergent and water. If the equipment is visibly soiled with blood or other body fluids or the patient is under contact precautions, it should be cleaned and disinfected before it is reused.
- **Floors**: Clean floors frequently (daily and as needed) with a wet mop, detergent and water. A disinfectant should be used when contamination is present, such as for blood or other body fluid spills as described below.
- **Sinks**: Scrub frequently (daily or more often as needed) with a separate mop, cloth or brush and a **disinfectant cleaning solution**. Rinse with water.
- **Toilets and latrines**: Scrub frequently (daily and more often as needed) with a separate mop, cloth or brush and a **disinfectant cleaning solution**.
- **Patient rooms**: Clean daily and after patient discharge, using the processes described above. The same cleaning process applies to rooms of patients who are under isolation precautions. Any cleaning equipment used in the rooms of patients under isolation precautions should be cleaned and disinfected before used in another room.
- **Procedure rooms**: Wipe horizontal surfaces, equipment and furniture used for the procedures with a **disinfectant cleaning solution** after each procedure and whenever visibly soiled. Clean blood or other body fluid spills as described below.
- **Examination rooms**: Wipe horizontal surfaces with a **disinfectant cleaning solution** after each procedure and whenever visibly soiled. Linen or paper on the examination table should be changed after each patient. Clean blood or other body fluid spills as described below.
- **Laboratory**: Wipe countertops with a **disinfectant cleaning solution** after each shift and whenever visibly soiled. Clean blood or other body fluid spills as described below.
- **Curtains**: Change and clean curtains according to the routine schedule and when visibly soiled.
- **Carpets**: Vacuum carpets daily in patient rooms, or weekly in offices or conference rooms.
- **Soiled linen**: Collect soiled linen daily (or more often as needed) in closed, leakproof containers.

- **Waste**: Collect waste from all areas at least daily (or more frequently as needed). **Avoid overflowing**.
- **Waste containers**: Clean contaminated waste containers after emptying each time. Clean noncontaminated waste containers when visibly soiled and at least once a week. Use a **disinfectant cleaning solution** and scrub to remove soil and organic material.

SCHEDULE AND PROCEDURES FOR THE OPERATING ROOM

- At the beginning of each day, all flat (horizontal) surfaces (table, chairs, etc.) should be wiped with a clean, lint-free moist cloth to remove dust and lint that may have collected overnight.
- Total cleaning is **not necessary** between each case for surgical procedures.
- Total cleaning or terminal cleaning (mopping floors and scrubbing all surfaces from top to bottom) of the operating room should be done at the **end** of each day.

Note: Do not dry mop or sweep the operating room. (This causes dust, debris and microorganisms to become airborne and contaminate clean surfaces.)

Total Cleaning

STEP 1: Move covered decontamination buckets to the central supply or processing room. A clean bucket containing a fresh 0.5% chlorine solution, or other locally available and approved disinfectant, should be provided at the beginning of each day and after each case.

Remember: All areas of the surgical suite, scrub sinks, scrub or utility areas, hallways and equipment should be totally cleaned, regardless of whether they were used during the 24-hour surgery period.

STEP 2: Remove covered contaminated waste container and replace it with a clean container. Arrange for burning (incineration) or burial as soon as possible.

STEP 3: Close and remove sharps containers when three quarters full.

STEP 4: Remove soiled linen in closed leakproof containers.

STEP 5: Soak a cloth in disinfectant cleaning solution and wipe down all surfaces, including counters, tabletops, sinks, lights, etc. Wash from top to bottom, so that any debris that falls on the floor will be cleaned up last.

Note: If walls and ceilings are deteriorating or damp, cover with clean plastic sheets during procedures.

- **Walls and ceilings**. Wipe with a damp cloth, detergent and water as needed for visible soil.
- **Chairs, lamps, sinks, tabletops and counters**. Wipe with a damp cloth and disinfectant cleaning solution.
- **Operating room lamp**. Wipe with a damp cloth and disinfectant cleaning solution.
- **Operating room table**. Wipe with a 0.5% chlorine solution (or other approved disinfectant) to decontaminate. Then clean top, sides, base, legs and any accessories (e.g., leg stirrups) with a damp cloth and disinfectant cleaning solution.
- **Floors**. Clean with a wet mop using a disinfectant cleaning solution.

Note: The double- or triple-bucket method is recommended for the cleaning of the operating room and other areas of the surgical suite.

- **Vents** (heating or air conditioning). Wipe with a damp cloth, soap and water.

Note: Cleaning the filters in air conditioners regularly will help them run more efficiently and decrease the growth of molds.

Between each case, do the following:

- **Spills**. Clean spills with a 0.5% chlorine solution or other locally available and approved disinfectant (see below).
- **Operating room bed**. Wipe all surfaces and mattress pads with a **disinfectant cleaning solution.**
- **Instrument tables (trolley and Mayo stand) and other flat surfaces**. Wipe all flat surfaces that have come in immediate contact with a patient or body fluids with a disinfectant cleaning solution.
- **Center of operating room surrounding the operating room bed**. Mop with a disinfectant cleaning solution (if visibly soiled).

Remember: Because all patients are considered potentially susceptible and infectious, Standard Precautions are used; no additional measures are necessary if a client is known to have an infection.

- **Waste**. Collect and remove all waste from the operating room in closed leakproof containers.
- **Sharps containers**. Close and remove containers from the operating room when they are three quarters full.
- **Containers with a 0.5% chlorine solution for decontamination**. Remove covered containers with instruments from the operating room and replace them with clean containers with a fresh 0.5% chlorine solution.
- **Soiled linen**. Remove soiled linen in leakproof, covered waste containers.

HOW TO CLEAN SPILLS OF BLOOD AND OTHER BODY FLUIDS

Clean spills of blood, body fluids and other potentially infectious fluids **immediately**:

- For **small spills**. While wearing utility or examination gloves, remove visible material using a cloth soaked in a 0.5% chlorine solution, then wipe clean with a disinfectant cleaning solution.
- For **large spills**. While wearing gloves, flood the area with a 0.5% chlorine solution, mop up the solution and then clean as usual with detergent and water.

HOW TO CLEAN SOILED AND CONTAMINATED CLEANING EQUIPMENT

STEP 1: Decontaminate cleaning equipment that has been contaminated with blood or body fluids by soaking it for 10 minutes in a 0.5% chlorine solution or other locally available and approved disinfectants.

STEP 2: Wash cleaning buckets, cloths, brushes and mops with detergent and water daily, or sooner if visibly dirty.

STEP 3: Rinse in clean water.

STEP 4: Dry completely before reuse. (Wet cloths and mop heads are heavily contaminated with microorganisms.)

REFERENCES

Arnow P et al. 1991. Endemic and epidemic aspergillosis associated with in-hospital replication of Aspergillus organisms. *J Infect Dis* 164(5): 998–1002.

Centers for Disease Control and Prevention (CDC). 1991. Chlorine gas toxicity from mixture of bleach with other cleaning products. *MMWR* 40(36): 619–621.

Centers for Disease Control (CDC). 1988. Guideline for handwashing and environmental control, 1985. *MMWR* 37(24).

Chou T. 2002. Environmental Services, in *APIC Text of Infection Control and Epidemiology*. Association for Professionals in Infection Control and Epidemiology (APIC): Washington, DC, pp 73–81.

McFarland LV et al. 1989. Nosocomial acquisition of *Clostridium difficile* infection. *New Engl J Med* 320(4): 204–210.

Russell AD, WB Hugo and GA Ayliffe. 1982. *Principles and Practice of Disinfection, Preservation and Sterilization.* Blackwell Scientific Publications: Oxford, England.

South East Asia Regional Office (SEARO), World Health Organization. 1988. *A Manual on Infection Control in Health Facilities*. SEARO: New Delhi, India, pp 72–88.

SEVENTEEN

CLINICAL LABORATORY SERVICES

KEY CONCEPTS you will learn in this chapter include:

- What the special exposure risks to laboratory staff are
- How exposures or accidental injuries occur
- How laboratories are classified according to exposure risk to staff, fellow workers and the environment
- What biosafety and infection prevention practices in clinical laboratories are

BACKGROUND

Any laboratory worker who handles blood or potentially infected body fluids is at some risk of accidental injury or exposure. But staff working in clinical laboratories or research units isolating or handling pathogenic microorganisms (e.g., vaccine development) are at the greatest risk. For workers in these areas, guidelines for various levels of risk (biosafety levels) have been developed. These guidelines are very specific and describe the appropriate containment equipment, facilities and procedures to be used by staff at all times.[1] For less risky areas, such as routine chemistry, clinical pathology and blood banks, general infection prevention guidelines and recommendations have been developed.

Prior to the emergence of HIV/AIDS and the re-emergence of multidrug-resistant tuberculosis in the 1990s, little progress in reducing laboratory-acquired infections was made (i.e., annual incidence of about 3 per 1000 laboratory workers in the US) (Grist and Emslie 1991). More recently, however, not only has there been renewed interest in biosafety efforts, but compliance among health workers may even be increasing. In developing countries, however, the situation is quite different. For example, in a recent report by Mujeeb et al (2003), of the 44 clinical laboratories in Karachi, Pakistan gloves were used in only two (4.5%) laboratories and disinfectants in seven (16%). Moreover, only seven laboratories (16%) had access to an incinerator.

The biosafety guidelines described in this chapter are designed for the prevention of laboratory-acquired infections in general hospital settings. They are aimed at containing the biohazardous agents and educating laboratory workers about the occupational risks. The recommendations

[1] Detailed information on recommendations for specific bacterial, fungal, parasitic and viral agents can be found on the CDC website at http://www.cdc.gov/od/ohs/biosfty/bmbl/sections7.htm

cover safe work practices, laboratory design, the use of appropriate personal protective equipment (PPE) and waste management. Adherence to these biosafety guidelines can reduce the risk of exposure and subsequent laboratory-acquired infections (Harding et al 1995).

DEFINITIONS

- **Biosafety level (BSL) guidelines**. Combination of primary and secondary containment and safety guidelines designed for use in microbiology laboratories and bacteriology research units functioning at four levels (BSL-1 to BSL-4) of increasing risk:

  - **BSL-1** is the lowest level of containment and microbiologic safety guidelines and is entirely based on standard laboratory practices. These guidelines are recommended for those working with microorganisms, such as *Bacillus subtilis*, that are not known to cause infections in healthy adults.
  - **BSL-2** is generally applied in bacteriology laboratories working with agents (e.g., *Salmonella* species) associated with human diseases of varying severity. When standard microbiologic practices are applied, the agents may be handled on open benches, especially if primary barriers, such as facemasks, gowns and examination gloves, are used when appropriate. The use of biologic safety cabinets (BSCs) and safety centrifuges may be necessary.
  - **BSL-3** is aimed at containing hazardous microorganisms primarily transmitted by the airborne route (aerosols and droplets), such as tuberculosis or varicella (chicken pox). Laboratory staff who work in these situations must be trained in the use of appropriate equipment, including suitable ventilation systems and the use of BSCs.
  - **BSL-4** is designed for use where agents causing life-threatening or untreatable diseases that can affect the laboratory worker via the airborne route are present, such as hemorrhagic fever viruses. Trained workers using level III BSCs or wearing full-body, air-supported positive pressure suits must perform all procedures in these laboratories. In addition, the facility itself must be totally isolated from other laboratories and have specialized ventilation and waste management systems.

- **Biological safety cabinets (BSCs)**. Devices that provide protection for personnel, the agent being processed and the environment. They range in complexity from level I (general research cabinets for use with low- to moderate-risk microorganisms) to level III (totally enclosed cabinets with gas-tight construction that provide maximum protection to workers and the environment).

Note: Gloves should be pulled over the cuffs of gowns to protect the wrists.

- **Laboratory-acquired infections**. Nosocomial infections resulting from the performance of laboratory activities by staff, regardless of how they occurred.

TYPES OF EXPOSURE RESULTING IN LABORATORY-ACQUIRED INFECTIONS

Infections from pathogenic organisms occur by several means. The most common are the following:

- **Inhalation**. Mixing, grinding or blending an infectious agent or flaming a transfer loop can generate aerosols that can be inhaled by unprotected workers.
- **Ingestion**. Workers may be exposed through:
  - unconscious hand-to-mouth actions;
  - placing contaminated articles (pencils) or fingers (when biting fingernails) in the mouth;
  - eating, drinking or smoking in the laboratory or failing to use proper hand hygiene (neglecting to wash hands or to use a waterless, alcohol-based antiseptic handrub before and after eating); or
  - pipetting (13% of accidental laboratory-acquired infections are associated with mouth pipetting).

Note: Sharps should be handled with care and disposed of immediately after use in sharps containers located close to the work area.

- **Puncture wounds**. Accidental injury with sharps (suture needles, scalpel blades and contaminated broken glassware) is the leading cause of laboratory-acquired infections.
- **Contamination of skin and mucous membranes**. Splashes and sprays of contaminated fluids onto mucous membranes of the mouth, nasal cavity and conjunctivae of the eyes, and hand-to-face actions can lead to the transmission of pathogenic organisms.

Note: Wearing a simple, plastic facemask or shield can minimize these risks.

BIOSAFETY AND RECOMMENDED INFECTION PREVENTION PRACTICES FOR LABORATORY WORKERS

Laboratory workers in hospitals and clinics handling blood, potentially contaminated body fluids or specimens containing pathogenic microorganisms need to be aware of the potential hazards of these infectious agents and materials and know how to protect themselves, fellow workers and the environment. Most hospital or clinic laboratories are defined as BSL-1 or BSL-2 units. As such, prevention of occupationally-acquired infections in these laboratories consists primarily of staff conscientiously using the basic practices described for all healthcare workers, namely, hand hygiene (handwashing or use of an antiseptic handrub) before and after eating or contact with infectious materials, and the use of protective gloves, facemasks and gowns as

appropriate (see **Chapters 2–6**). Because the infectious agents they may encounter are classified as low or moderate risk, special containment practices are not required (i.e., these agents are not a significant risk to the environment and can be disposed of as any other infectious hospital waste as described in **Chapter 8**).

For staff working in bacteriology laboratories or microbiologic research units (BSL-3 or 4), containment of hazardous agents to protect the environment is an added requirement for the safe handling of these infectious agents. As described above, depending on the type of organisms being handled, BSCs and other PPE (e.g., full-body, air-supported positive pressure suits) are required and staff must be fully trained in their use.

General Biosafety and Infection Prevention Guidelines

- Wear new examination gloves when handling blood, body fluids and/or specimens containing pathogenic microorganisms.
- No eating, drinking or smoking is permitted in the laboratory.
- Food should not be stored in refrigerators used for clinical or research specimens.
- No mouth pipetting is permitted; use proper mechanical devices (e.g., suction bulbs).
- Do not open centrifuges while still in motion.
- Always cover the end of blood collection tubes with a cloth or paper towel, or point them away from anyone's face when opening.
- Decontaminate work surfaces daily or when contaminated, such as after spills, with a 0.5% chlorine solution.
- Wear protective face shields or masks and goggles if splashes and sprays of blood, body fluids, or fluids containing infectious agents are possible.
- Wear heavy-duty or utility gloves when cleaning laboratory glassware.
- Use puncture-resistant, leakproof containers for sharps.
- Place infectious waste materials in plastic bags or containers.

Blood Drawing (Phlebotomy)

CDC considers blood drawing (phlebotomy) to be one of the highest-risk sharps procedures. This is because the most frequently used needles are large bore (18 to 22 gauge), and a considerable amount of blood is left in the needle after use.[2] In a 1999 report (EPINet), 21% of 1,993 sharps injuries reported in the US were associated with blood drawing (venous or arterial blood samples and finger/heelsticks). Over 80% of the needlesticks occurred when drawing venous blood, using either a vacuum-tube blood collection needle, disposable needle and syringe or butterfly needle.

When collecting a blood specimen (phlebotomy) be sure to:

[2] HIV can survive in needles and syringes for more than 4 weeks at room temperature (Abdala et al 1999; Rich et al 1998).

- Wear examination gloves.
- Have assistance when patients might be uncooperative (children, mentally impaired, etc.).
- Have assistance for holding children when doing heel sticks.

REFERENCES

Abdala N et al. 1999. Survival of HIV-1 in syringes: Effects of temperature during storage. *J Acquir Immune Defic Syndr Hum Retrovirol* 20(1): 73–80.

Exposure Prevention Information Network (EPINet) Data Reports. 1999. *Uniform Needlestick and Sharp Object Injury Report 21 Hospitals, 1999*. International Healthcare Worker Safety Center, University of Virginia. Available on: www.med.virgina.edu/epinet/soi99.html.

Grist NR and JA Emslie. 1991. Infections in British clinical laboratories, 1988–1989. *J Clin Pathol* 44(8): 667–669.

Harding L et al. (eds). 1995. *Laboratory Safety: Principles and Practices*, 2nd ed. American Society for Microbiology: Washington, DC.

Mujeeb SA et al. 2003. Infection control practices in clinical laboratories in Pakistan. *Infect Control Hosp Epidemiol* 24(2): 141–142.

Rich JD et al. 1998. Detection of HIV-1 nucleic acid and HIV-1 antibodies in needles and syringes used for non-intravenous injection. *AIDS* 12(17): 2345–2350.

EIGHTEEN

BLOOD BANK AND TRANSFUSION SERVICES

KEY CONCEPTS you will learn in this chapter include:

- Why provision of transfusions is unsafe in many settings
- What the special risks to patients receiving transfusions are
- What the indicators for transfusion are
- How complications and the risk of infection from transfusions can be prevented

BACKGROUND

Blood bank and transfusion services collect, process, store and provide human blood intended for transfusion, perform pretransfusion testing and, finally, infusion into a patient. Although these processes may take place in a single hospital department, often they are performed in two separate places. For example, in many countries most blood for transfusion is collected in blood centers, which then process, store and transport it for use by a hospital's transfusion service. The transfusion service in turn is responsible for maintaining an adequate supply of needed blood and blood products, blood-typing and cross-matching patients, and releasing the blood for transfusion.

In many respects, the infusion of blood or blood products is equivalent to the use of any other intravenous therapeutic agent (e.g., antibiotics). There are, however, additional, specific risks to patients receiving transfusions. For example, because of the potential risk of patients receiving transfusions being exposed to serious infections (HBV, HCV or HIV), guidelines for competently and safely performing various screening and testing processes and procedures have been developed. These guidelines are very specific, allow little room for variation in practice and need to be followed by staff at all times if transfusion services are to be safely provided.[1] As a consequence, in developed countries, blood bank and transfusion services are highly regulated, and the quality of services is monitored daily (AABB 2002).

Staff working in blood banks and transfusion services also are at risk of accidental injury (e.g., needlestick) or exposure to contaminated blood or blood products. To protect themselves, staff need to know and understand the importance of handwashing, use of gloves and personal protective

[1] Although there are a number of books that deal with transfusion, the *Standards for Blood Banks and Transfusion Services,* prepared by the American Association of Blood Banks (AABB 2002), is the only manual that provides uniform codes of practice for use in the United States.

equipment such as face shields or masks and plastic aprons, where appropriate.

In this chapter, the guidelines for the safe provision of blood bank and transfusion services are summarized from the perspective of:

- screening the blood donor,
- ensuring the safety of the donor,
- testing to make sure the blood or blood product is safe for use,
- protecting the patient receiving the transfusion, and
- ensuring the safety of laboratory and clinical staff.

Adherence to these guidelines can reduce the risk of transfusion-related complications and hospital-acquired (nosocomial) infections in patients and exposure to and subsequent laboratory-acquired infections in staff (Harding et al 1995).

DEFINITIONS

- **Blood bank**. Facility or hospital unit that performs the collection, processing, storage and distribution of human blood or blood products.
- **Clinically significant antibody**. An antibody capable of producing an adverse reaction to transfused blood or blood product obtained from a donor (allogenic antibody) or recipient (autologous antibody).
- **Closed system for obtaining blood**. System in which the blood is not exposed to air or outside elements during collection and processing, including separation of components (e.g., platelets) if required prior to transfusion. It is the safest way to collect, process and store blood.
- **Donor-Patient**. Person whose blood is collected for possible transfusion to another person (allogenic transfusion).
- **Donor-Recipient**. Person whose own blood is collected for possible transfusion to her/himself (autologous transfusion).
- **Lookback system**. Process of identifying persons who have received blood transfusions from donors who are subsequently found to have infections with HCV, HIV (and often HBV), and notifying them if appropriate.
- **Recipient transfusion reaction**. Adverse reaction to infusing blood or blood products into a patient (recipient). It may occur at any time during the transfusion but often happens shortly after starting it. The reaction may be mild or severe and rarely is fatal. Types of reactions include allergic (from mild itching and hives to serious breathing problems) and hemolytic (destruction of red cells) reactions as well as fever, chills, rapid heart rate (tachycardia), hyperventilation, fainting and, rarely, cardiac arrest. Delayed reactions several days or weeks

after the transfusion may occur and may be due to serum sickness (antigen-antibody reaction).

- **Transfusion service**. Facility or hospital unit that provides storage, pretransfusion testing and cross-matching, and infusion of blood or blood products to intended patients (recipients).
- **Unit of blood**. Sterile plastic bag in which a fixed volume of blood is collected in a suitable amount of anticoagulant. (The collection system should be a closed system, usually consisting of a sterile hypodermic needle connected by tubing to a collection bag or bottle that has one or two sterile ports for inserting a sterile blood administration set.)
- **Urticarial reaction**. Allergic reaction consisting of itching (pruritis), hives, skin rash and/or similar allergic condition occurring during or following a transfusion of blood or blood products.

WHY TRANSFUSION SERVICES ARE UNSAFE IN MANY SETTINGS

Transfusing patients with blood and blood products is one the oldest medical and surgical remedies. In resource-poor countries, it is one of the few procedures available to practitioners. As a result, it is overused and provided for a myriad of reasons, many of which are not appropriate. Moreover, all too often blood is obtained from paid, high-risk donors such as commercial sex workers and intravenous drug users who are minimally screened for infectious diseases or other conditions (e.g., anemia) that normally should disqualify them as donors. For example, it is estimated that less than half of the world's blood supply used for transfusions is safe.

In addition, staff working in these units, as well as health workers giving the transfusions, often have received little training and are not aware of the risks to their patients and themselves. As a consequence, even if rapid test kits for infectious disease testing are available, staff working in the blood bank or transfusion service may not know how to use them or interpret the results. In addition, a common problem expressed by many transfusion service staff is that physicians demand release of the blood before testing is completed even in nonemergency situations—and in some emergency situations even before cross-matching is done. Because most transfusion services lack lookback record keeping capability, patients who are transfused with blood or blood products that are subsequently found to be seropositive for HBV, HCV or HIV are seldom notified.

PROVISION OF SERVICES

Blood bank and transfusion services involve:

- selecting donors and assuring that they are informed;
- collecting blood from screened donors;
- testing for blood components, antibodies and infectious diseases;

- storing and transporting blood;
- pretransfusion testing of patient (recipient) blood; and
- transfusing patients.

Donor Selection and Informed Consent

To attract volunteer donors and encourage their continued participation, the place where blood is collected should be kept clean and be as pleasant, safe and convenient as possible.

Donor Selection

The donor selection process is one of the most important steps in protecting the safety of the blood supply. The process is intended to identify medical problems, behaviors (e.g., IV drug use) or events that put a person at risk of being infected and transmitting a serious disease to the person receiving the transfusion. To accomplish this, donors should be questioned about their medical history, be given a limited physical examination (e.g., pulse and blood pressure checked and heart and lungs listened to with a stethoscope) and have their hemoglobin or hematocrit determined. In general, potential donors should be at least 17 years old, unless there are special circumstances requiring a minor to give blood. They should be in good health, not severely anemic (Hg > 11 g/dL or Hct > 33%) and not be infected (carrier or seropositive) for HBV (if not vaccinated), HCV or HIV/AIDS.[2]

Informed Consent

Prior to collection of blood, the elements of the donation process should be explained in simple, easily understandable terms using the patient's primary language if possible. The explanation should include information about the risks of venipuncture (phlebitis or local infection and rarely bacteremia or septicemia) and potential adverse responses to having 400–500 mL of blood removed (tachycardia, hyperventilation, feeling light-headed and occasionally fainting). It also should include mention of the tests performed to reduce the risk of transmission of infectious diseases, such as syphilis and serious bloodborne viruses, to patients (recipients). In addition, the donor should have an opportunity to ask questions about the procedure and to refuse consent. In particular, the donor should clearly understand exactly how donors will be notified about any medically important abnormality found during the predonation evaluation or as a result of laboratory testing (e.g., a positive rapid test for HIV) and followup. Appropriate education, counseling and referral should be offered as well.

[2] For the most current Uniform Donor History Questionnaire, check the AABB website (www.aabb.org).

Blood Collection

The donor as well as the future recipient should be protected by proper collection of the blood. Careful skin preparation using an aseptic technique is a critical component of donor and recipient safety. Several studies suggest that fewer than two or three blood units per thousand will contain bacteria if aseptic technique is used and blood is collected in a closed system (Abrutyn, Goldman and Scheckler 1998). To minimize the risk of contamination:

STEP 1: Make sure all items are available:

- Blood collection set (closed system) consisting of a sterile plastic bag containing a sufficient amount of anticoagulant for the quantity of blood to be collected, IV tubing and large-gauge #18 or #19 hypodermic needle)
- Pair of sterile or high-level disinfected surgical gloves
- Clean tourniquet or blood pressure cuff
- Antiseptic solution (e.g., 2% chlorhexidine gluconate, 60–90% alcohol or 10% povidone iodine) and sterile or clean gauze squares (2 x 2) or cotton swabs
- Surgical tape
- Towel to place under patient's hand or forearm
- Basin of clean warm water, soap, face cloth and clean dry towel to wash it.[3] (Only needed if patient's arm is visibly soiled.)
- Plastic bag or leakproof, covered waste container for disposal of contaminated items
- Puncture-resistant sharps containers (within arm's reach if possible)

Note: In 1991, Maki, Ringer and Alvarado reported that the infection rate with chlorhexidine was 84% lower than with povidone-iodine (PVI) or alcohol for skin antisepsis.

STEP 2: Explain procedure to the patient.

STEP 3: Identify the best veins for inserting the IV needle. (Blood should be drawn from a large, firm vein—usually the antecubital space—that is free of skin lesions or rashes. Both arms should be checked.)

STEP 4: Put the tourniquet or blood pressure cuff on the upper arm about 9 cm (3–4 inches) above the antecubital space to confirm that the vein is visible and then release the tourniquet or cuff.

STEP 5: If the venipuncture site is visibly soiled, first wash it with soap and clean water and dry with a clean cloth.[3]

STEP 6: Wash hands with soap and clean water and dry with a clean, dry towel or air dry.[3] (Alternatively, if hands are clean, apply 5 mL,

[3] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

about 1 teaspoonful, of a waterless, alcohol-based antiseptic handrub to both hands and vigorously rub hands and between fingers until dry.)

STEP 7: Place the donor's arm on a clean towel and cleanse an area about 3 cm (1.5 inches) in diameter with antiseptic solution. Use a circular motion outward from the proposed needle insertion site over the vein.

If using polyvidone-iodine or other iodophor, allow it to dry (about 2 minutes) because PVI only releases free iodine, the active antiseptic agent, slowly (see **Chapter 6**).

STEP 8: Do not touch the area after applying the antiseptic solution.

STEP 9: Put the tourniquet or blood pressure cuff on the upper arm again.

STEP 10: Put sterile or high-level disinfected gloves on both hands.

STEP 11: Insert the hypodermic needle into the vein without touching the skin if possible, release the tourniquet or cuff and then secure the needle by placing a short piece of tape across the blood collection tubing below the area cleansed with antiseptic.

Note: If a blood pressure cuff is used, collect the blood under about 40 to 60 mm Hg pressure, but if a tourniquet is used it should be applied just tight enough to keep the vein full and firm, but not so tight as to cause discomfort to the donor.

STEP 12: When the required amount of blood has been obtained, remove the needle without touching the barrel or tip of the needle and place it in a puncture-resistant sharps container.

STEP 13: Cover the insertion site with a 2 x 2 gauze square, and apply pressure until the bleeding stops. (The donor can be shown how to apply pressure as it may take several minutes before all bleeding stops.)

STEP 14: Check the arm. If the bleeding has stopped, secure the gauze squares using 1 or 2 short pieces of surgical tape.

STEP 15: Prior to removing gloves, place any blood-contaminated waste items (cotton or gauze squares) in a plastic bag or leakproof, covered waste container.

STEP 16: Remove gloves by inverting and place them either in a plastic bag or waste container.

STEP 17: Wash hands or use an antiseptic handrub as above.

STEP 18: Have the patient remain resting on a bed or in the donor chair for several minutes.

STEP 19: Provide the donor with something to drink and a piece of bread, a cookie or a biscuit.

STEP 20: Tell the donor to drink more fluids during the next 4 hours and avoid alcohol or smoking until more food has been eaten. Also, tell her/him that if there is bleeding apply pressure and raise the arm over the head. Finally, if the donor becomes dizzy or sick to the stomach

(nauseated), tell her/him to sit down, bend forward and rest her/his head between the knees until the dizziness or nausea passes.[4]

Once the blood is collected, contamination can be avoided by:

- maintaining appropriate storage conditions,
- testing the blood unit without entering the closed collection system, and
- infusing or discarding the blood unit within a short period once the closed system has been opened (AABB 2002).

Blood Component and Infectious Disease Testing

The tests generally required to be performed on all blood or blood products that are intended for transfusion to patients include the following:

- ABO blood group is determined by testing the donor's red cells with anti-A and anti-B reagents and by testing the donor's serum or plasma A_1 and B red cells.
- Rh type is determined by testing with anti-D reagent. If the initial test with anti-D is negative, the blood should be additionally tested using a method designed to detect weak D.
- Blood from donors with a history of transfusions or pregnancy should be tested for unexpected antibodies to red cell antibodies using methods to demonstrate clinically significant antibodies.

Note: When either test is positive, the blood unit shall be labeled as Rh POSITIVE. If both tests are negative, then it is labeled as Rh NEGATIVE.

In addition, donor blood should be tested for several infectious diseases. Blood should not be released for transfusion unless the results are negative for all tests, with the exception of the test for syphilis that has been shown to be a biologic false positive.[5] The recommended tests include:

- syphilis by screening with a standard antibody test such as the rapid plasma reagent (RPR) test,
- hepatitis B virus by testing for the hepatitis B surface antigen (HbsAg) and HBV core antigen (anti-HBc),
- hepatitis C virus by testing for anti-HCV, and
- human immunodeficiency virus by testing for type 1(HIV-1) antigen and antibodies to HIV-1 and HIV-2 antigens.[6]

[4] Rarely, donors may have convulsions or experience irregular or rapid heartbeats—occasionally even cardiac arrest.
[5] Persons with untreated syphilis most often have antibodies that can be detected by tests such as the RPR, but false positive antibodies can also develop, usually lasting for only a few weeks after bacterial or viral infections or after immunization procedures. In some patients with autoimmune diseases, especially lupus, these false positive antibodies persist indefinitely.
[6] A combination test for anti-HIV-1/2 may be used.

Blood Storage and Short Distance Transport

Blood units must be stored in a refrigerator that can be maintained at temperatures between 1–6°C (34–46°F). There must be a system to monitor temperatures continuously and record them at least every 4 hours.[7] In addition, the refrigerator should have an alarm system that signals by sound before the blood reaches unacceptable storage temperatures.

Blood units exposed to a temperature above the accepted level for an unknown period should be discarded. To do this:

Remember: After removing gloves, wash hands or use a waterless, antiseptic handrub.

- Wear examination or utility gloves and protective eyewear.
- Pour contents down a utility sink drain, into a flushable toilet or latrine.
- Place empty blood bags and tubing in a plastic bag or leakproof, covered waste container.
- Dispose of plastic bags or contents of the container according to hospital or facility waste management guidelines.

Blood units transported short distances (e.g., from the blood bank or transfusion service to the ward or operating room) require no special handling. Blood should not, however, be allowed to reach temperatures outside the acceptable range.

Pretransfusion Testing and Cross-matching

The purpose of pretransfusion testing is to select blood or blood products that will not cause harm to the patient (recipient), and to ensure that the red cells will survive (not be destroyed too rapidly) when transfused. When performed properly, pretransfusion tests will confirm the ABO group of the red cells, Rh status, the presence of clinically significant red cell antibodies in the recipient's blood and compatibility between selected samples of donor blood with the recipient's blood (cross-matching).

Note: If a discrepancy in ABO group is detected and transfusion is necessary before the problem can be solved, only group O red cells should be used.

The **first step** is to test a sample of recipient blood using the same methods and recommended infection prevention practices used to test donor blood.[8] (Recipient plasma or serum, however, need only be tested with anti-D reagent, as the test for weak anti-D is not necessary.)

Note: For repeat testing, only anti-D reagent needs to be used.

To avoid the 80% chance of Rh sensitization, Rh-positive blood should not be given to a patient who is Rh negative. Occasionally, however, ABO compatible Rh-negative blood is not available. In this case, the alternatives are either to postpone the transfusion until Rh-negative ABO compatible blood is available or, if circumstances warrant, to give Rh-positive blood. If the patient is a woman, and depending on her childbearing potential and

[7] A thermometer placed inside the refrigerator and checked at regular intervals can be used if an automated system for monitoring and recording the interior temperature is not available or not working.

[8] It is acceptable to take the blood sample for blood typing and cross-match from an existing IV line if the patient has one in place.

the volume of the blood transfused, it may be desirable to give Rh immune globulin within 24 hours of the transfusion of Rh-positive blood.

The **second step** is repeat testing of the donor blood to confirm the ABO group and Rh.

Note: A negative red cell antibody screen does not guarantee that the recipient's serum is free of clinically significant antibodies, however, or that there will be normal survival of the red cells transfused.

The **third and final step** is to crossmatch the red cells of selected donor(s) against the serum or plasma of the recipient to be sure there are no ABO and clinically significant antibody problems. If antibodies are detected in the recipient's blood, the number or type of tests needed to ensure compatibility varies from case to case. (Most samples tested, however, have a negative screen for antibodies and are crossmatch-compatible with all selected donor units of blood.)

If no clinically significant antibodies were detected in the recipient's blood, and there is no prior record of antibodies, serologic cross-matching, which is quicker and less difficult to perform, is acceptable.

Note: If the patient's ABO group and Rh status are unknown, use O-negative blood, especially if the patient is a woman of childbearing age.

When blood is urgently needed, the physician must decide whether to transfuse with uncrossmatched or partially crossmatched blood or to delay the transfusion. The risk of transfusing blood without cross-matching is that a serious reaction may occur, including the rapid breakdown (hemolysis) of transfused red cells.

TRANSFUSION OF BLOOD OR BLOOD COMPONENTS

Like any other medical treatment, the decision to transfuse a patient should be based on the need (indications) for transfusion of blood or blood components in comparison with the risks, potential benefits and alternatives. In addition, before receiving a transfusion, the patient should be told the reason(s) for needing a transfusion, clearly understand and accept the risks and have any questions answered regarding the procedure. (If the patient is unconscious or incapable of giving consent, when possible a spouse, relative or adult friend should give consent.)

Indications

The main reason for transfusion of whole blood or packed red blood cells is to increase the oxygen-carrying capacity to meet the tissue demands for oxygen. The two primary conditions are:

Note: In situations of acute bleeding, the transfusion threshold is 30–40% blood loss for otherwise healthy adults, provided blood volume is maintained (ASATF 1996).[9]

1. actively bleeding patients (acute blood loss), and
2. patients with chronic or symptomatic anemia.

For the former, the objectives of initial treatment are to stop the bleeding and to restore intravascular volume in order to prevent hypovolemic shock (shock due to decreased fluid in the circulation). Thus, the immediate need

[9] If the blood volume is maintained, healthy resting adults are able to tolerate an acute decrease in red cells to a hemoglobin of 5 g/dL without evidence of lack of tissue oxygenation (Weiskopf et al 1998).

is to give IV fluids that will help restore the circulation, and then restore oxygen-carrying capacity.

> The generally accepted hemoglobin level for transfusing patients with acute blood loss is 7 g/dL, with those patients having a level of 6 g/dL almost always requiring transfusion but those with a level of 10 g/dL rarely needing it (ASATF 1996).

For chronic anemia, the objective should be to prevent patients from being symptomatic—weakness, dizziness, breathlessness, heart palpitations or rapid heart rate (Hebert 1999). Generally this means keeping the hemoglobin levels between 7 and 9 g/dL.

Transfusing Patients

Transfusion with donor whole blood (allogenic transfusion), which provides red cells to increase oxygen-carrying capacity, has stable coagulation factors and contains plasma to expand blood volume, is seldom done anymore in the US and other developed countries because there are more reactions with whole blood than with blood products. Thus, for most cases of active bleeding (acute blood loss), packed red cells (plasma removed) plus volume-expanding IV fluids have become the standard. In countries with limited resources, however, whole blood is still the standard, except in large hospitals or referral centers. In a typical adult, one unit of whole blood or packed red cells will raise the hemoglobin about 1 g/dL, or the hematocrit about 3%.

Note: Transfusion of packed red cells increases oxygen-carrying capacity with less expansion of blood volume per unit. This can be important in patients who are at risk of volume overload (e.g., newborns and patients with congestive heart failure).

Before starting the transfusion:

Note: Patients who have had blood transfusion reactions may have greater fear of transfusion, and certain types of reactions may increase the chance of recurrence.

STEP 1: Explain the procedure to the patient, determine if s/he has ever had a transfusion and record adverse reactions, if any.

STEP 2: With another health worker, correctly identify the blood product and patient:

- Confirm patient's name and check armband if available.
- Check compatibility tag attached to blood bag, including date blood is out of date and should not be used.
- For whole blood, check ABO group and Rh type, which should be on the patient's chart.
- Double check blood or type of blood product with the physician's order.
- Check blood for clots.
- Record baseline pulse and blood pressure.

Note: With the exception of sterile isotonic (0.9%) saline, **no drugs or medications should be added to whole blood units or blood components**.

Note: If a reaction is suspected, **stop** the transfusion, flush the line with sterile isotonic saline solution and infuse slowly to keep the IV line open, and notify the blood bank or transfusion service and physician.

STEP 3: Ask the patient to report chills, headaches, itching or rash immediately.

(The detailed steps for starting a peripheral intravenous line with a large-gauge needle or plastic catheter [#18 or #19] and setting up the blood administration set are described in **Chapter 24**.)

STEP 4: Once the transfusion has begun:

- take the patient's pulse and blood pressure every 5 minutes for the first 15 minutes of transfusion, and hourly thereafter.
- Observe the patient for flushing (red face or cheeks), itching, difficulty breathing, hives (clear fluid filled lesions on the skin) or other rash when checking vital signs.

STEP 5: When the transfusion is completed, record administration of the blood or blood product in patient's chart.

(The detailed steps for removing and disposing of the blood administration set, IV tubing and needle as well as any blood-contaminated waste items, are described in **Chapter 24**.)

PREVENTING COMPLICATIONS AND NOSOCOMIAL INFECTIONS

The collection, processing, storage and transfusion of blood and blood products is an essential service that all acute care hospitals and ambulatory surgical centers must be prepared to provide with high standards of quality. In addition, the safety of blood donors, patients (transfusion recipients), health workers and fellow staff requires that blood bank and transfusion service staff are qualified to perform the required tasks and follow recommended infection prevention practices consistently.

Preventing complications and nosocomial infections in patients requires that:

- Unnecessary transfusions are not given.
- Potential donors are adequately screened to minimize the risk of transmitting a serious bloodborne infection (e.g., syphilis, HBV, HCV and HIV).
- Donor blood is collected aseptically into a closed system to minimize contamination, and all steps in processing the blood are accomplished within this closed system.
- Prior to use, the blood or blood products are stored at the correct temperature and the expiration date has not expired prior to transfusion.
- All steps are taken to ensure that donor and patient blood are compatible in terms of ABO grouping and Rh and that unexpected

clinically significant antibodies in the donor's or patient's blood have been identified.

- Prior to transfusion, all information matching the blood with the intended recipient has been verified to prevent possible mistakes that could harm the patient.
- Aseptic technique is used to establish the peripheral IV line for giving the transfusion.
- During the transfusion, the patient's vital signs are monitored and s/he is checked regularly for any adverse reaction.
- If an adverse reaction is thought to have occurred, the transfusion is stopped immediately, and the patient treated for any complications (e.g., fainting, convulsions, breathing or heart problems) and her/his blood is checked for hemolysis.

Protecting Healthcare Workers

Health staff working in blood banks and transfusion services are at risk of exposure to pathogenic organisms in blood in a number of ways. The most common are:

Note: Wear gloves when collecting and transfusing blood and performing various tests on blood.

- Exposure to blood while collecting the donor specimen, during testing and when infusing the blood.
- Accidental injury with sharps (needles, scalpel blades and contaminated broken glassware), the leading cause of laboratory-acquired infections.
- Splashes and sprays of blood onto mucous membranes of the mouth, nasal cavity and conjunctivae of the eyes. (Wearing a clear plastic facemask or shield, or a surgical mask and goggles, can minimize these risks.)

Note: Sharps should be handled with care and disposed of immediately after use in puncture-resistant sharps containers located close to the work area.

In addition, decontaminate work surfaces with 0.5% chlorine solution daily or when contaminated, such as after blood spills, and place infectious waste materials in plastic bags or leakproof, covered waste containers.

MAKING BLOOD BANK AND TRANSFUSION SERVICES BETTER AND SAFER

In countries where resources are limited, blood bank and transfusion services frequently are not supervised and poorly monitored, screening of prospective donors is limited, and testing for infectious diseases, even for syphilis, may not be available. Under these circumstances, complications (transfusion reactions) and transmission of life-threatening infections to unsuspecting patients occur all too often. Without the commitment and full support of ministry of health officials, hospital administrators and infection prevention committees or working groups to implement basic blood bank and transfusion service policies and guidelines, improving the quality and safety of blood transfusions is unlikely.

As outlined above, many of the processes and procedures that can improve the quality of blood bank and transfusion services and make then safer for patients, health workers and their fellow staff are inexpensive and doable. Improving performance and compliance with recommended policies and guidelines can be significantly enhanced if:

- There is consistent support by hospital administrators to improve the quality of services (e.g., identified deficiencies are corrected, dangerous practices eliminated and staff are actively encouraged to seek inexpensive solutions).
- Supervisors regularly provide feedback and reward appropriate behavior (e.g., better screening of donors, use of aseptic techniques when collecting specimens, handwashing after removal of gloves).
- Role models, especially physicians and other senior staff and faculty, actively support recommended policies and guidelines (Lipscomb and Rosenstock 1997).

REFERENCES

Abrutyn E, DA Goldman and WE Scheckler (eds). 1998. Transfusion services, in *Saunders Infection Control Reference Service*. WB Saunders Company: Philadelphia, pp 583–587.

American Association of Blood Banks (AABB). 2002. *Standards For Blood Banks and Transfusion Services,* 21st ed. American Association of Blood Banks: Bethesda, MD.

American Society of Anesthesiologists Task Force (ASATF). 1996. Practice guidelines for blood component therapy. *Anesthesiology* 84(3): 732–747.

Harding L et al (eds). 1995. *Laboratory Safety: Principles and Practices*, 2nd ed. American Society for Microbiology: Washington, DC.

Hebert PC et al. 1999. A multicenter, randomized, controlled clinical trial of transfusion requirements in critical care. *N Engl J Med* 340(6): 409–417.

Lipscomb J and R Rosenstock. 1997. Health care workers: protecting those who protect our health. *Infec Control Hosp Epidemiol* 18(6): 397–399.

Maki DG, M Ringer and CJ Alvarado. 1991. Prospective randomized trial of povidone-iodine, alcohol and chlorhexidine for prevention of infection associated with central venous and arterial catheters. *Lancet* 338(8763): 339–343.

Weiskopf RB et al. 1998. Human cardiovascular and metabolic response to acute, severe isovolemic anemia. *JAMA* 279(3): 217–221.

NINETEEN

MANAGEMENT OF AN INFECTION PREVENTION PROGRAM

KEY CONCEPTS you will learn in this chapter include:

- What the organizing principles for managing infection prevention programs are
- Who should be involved in managing the program
- What the purpose of the infection prevention working group is
- What the decision-making process involves
- What types of issues and problems are commonly encountered

BACKGROUND

Successful programs for preventing the spread of infectious diseases by any route (blood, body fluids, air, droplet or contact) in healthcare facilities are based on understanding the scope of the problem, prioritizing activities and effectively using available resources. Because available resources are invariably limited, careful planning, implementing and monitoring activities on a regular basis, whether in a small clinic or a busy district hospital, are all essential.

In many countries, functioning infection surveillance systems are lacking, laboratory backup to identify the cause of nosocomial (hospital-acquired) infections is inadequate, and treatment options are limited. Thus, not only is infection prevention the most cost-effective option, but often it is the only realistic one available to limit the spread of disease within healthcare facilities. Unfortunately, it is the hardest of the three elements (surveillance, control and prevention) to implement because it requires staff at all levels to take an active role in preventing the spread of infections to patients, fellow workers and themselves.

Fortunately, most nosocomial infections in healthcare facilities can be prevented with readily available, relatively inexpensive strategies. And for some of the most serious infections, namely, AIDS, hepatitis C and multidrug-resistant tuberculosis, prevention is all we can do. To make this happen, however, healthcare administrators, clinic managers and staff at all levels must be totally committed to supporting and using recommended infection prevention guidelines and practices.

DEVELOPING SUCCESSFUL PROGRAMS

Helping hospitals and clinics become safer places in which to work or be cared for is largely about changing behavior. Education is not enough. To change unsatisfactory performance by staff (e.g., lack of compliance with handwashing guidelines) requires management reinforcement if the behavior change is going to be sustained (Lynch et al 1997). It is the responsibility of administrators and clinic managers, working in conjunction with key staff serving on operating room safety or infection prevention committees, to:

- set standards for performance, mentor staff and regularly monitor staff performance; and
- help staff at all levels "buy in" to using common sense when performing their assigned duties, as well as using appropriate personal protective equipment at all times.

In addition, there needs to be:

- Consistent support by hospital administrators and managers of safety efforts (e.g., identified deficiencies corrected, dangerous practices eliminated and staff actively encouraged to seek inexpensive, doable solutions).
- Supervisors who regularly provide feedback and reward appropriate behavior (e.g., handwashing between patient contacts).
- Role models, especially physicians and other senior staff and faculty, who actively support recommended infection prevention practices and demonstrate appropriate behavior (Lipscomb and Rosenstock 1997).

ORGANIZING PRINCIPLES FOR MANAGING INFECTION PREVENTION PROGRAMS

According to Lynch et al (1997), the three organizing principles for managing programs are:

1. establishing the relative importance of problems using the Spaulding categories of potential infection risk—critical, semicritical and noncritical;
2. identifying and analyzing the reasons for poor or incorrect performance; and
3. costing the issues (i.e., estimating the cost and benefits of activities).

As presented in **Chapter 1,** the Spaulding categories of potential risk provide a good basis for determining relative importance and setting priorities (e.g., the most serious and frequent problems and infections involve management in the critical area and, therefore, deserve the most attention and resources). The second principle, correctly identifying why

performance is not up to standard, usually comes down to three possible reasons. Staff:

1. do not know how to do the task correctly, or why they need to do it;
2. do not have the correct (adequate) protective equipment; or
3. lack motivation.

In most cases, more than one reason is involved. Understanding how these reasons contribute to performance deficits increases the potential for corrective action to be successful. The third and final principle is estimating the cost-benefit of corrective actions. In many countries, this is the most difficult of the three to implement because data on which to base estimates are often lacking.

WHO SHOULD BE INVOLVED IN MANAGING THE PROGRAM

As mentioned above, it is important to identify and bring together key hospital staff to form an infection prevention working group or committee if one has not been established. The purpose of the working group is to guide and support the use of recommended practices and to review and resolve related problems that may arise. This working group or committee should include representatives from a variety of patient care areas (e.g., surgery, central services, housekeeping, laboratory, purchasing and administration) and include one or more health professionals. In clinics where these functions often overlap, however, the group may consist of only two or three individuals.

Although the risk of infection cannot be completely eliminated, it can be minimized. Based on an analysis of the problems or issues, the working group will need to make and implement recommendations that are consistent with the relative importance, type of corrective action needed and cost.

Basic guidelines and activities that help managers implement successful programs include:

- Have written policies and procedures established to handle situations in which patients or staff are exposed to the risk of infection.
- Conduct staff orientation before new policies, recommendations or procedures are started and provide followup training when management reinforcement is needed.
- Be sure adequate supplies, equipment and facilities are available before start-up to ensure compliance.
- Conduct regular reviews to ensure the adequacy of the recommended changes or practices, to solve any new problems and to address staff concerns.

Remember: Include all staff members in what you are doing, share ideas and materials with them and be ready to listen to their points of view.

Finally, effective and regular communication at all levels is the key to developing the support needed for a successful program.

MAKING MANAGEMENT DECISIONS

With infection prevention, as with any clinical area, numerous situations arise where tough decisions have to be made, weighing the advantages of a certain procedure against the possible risks to the patient or healthcare worker. These decisions must be practical and consistent and, as much as possible, should be based on scientific evidence. Throughout this manual, evidence is presented to help managers make better, more informed decisions and recommendations regarding frequently encountered problems, such as:

- Recommendations for improving compliance with hand hygiene guidelines (**Chapter 2**).
- Appropriate selection and use of gloves for various healthcare tasks (**Chapter 4**).
- Selecting the most appropriate antiseptic agents or chemical disinfectants, ones that are affordable and usually locally available (**Chapters 6** and **12** and **Appendixes B** and **E**).
- Decisions regarding the appropriate reuse of disposable (single-use) items (**Chapter 14**).
- Use of personal protective equipment (PPE), especially gloves and other items (**Chapters 4** and **5**). (These items should be provided based on available resources and be made available in areas of the healthcare facility where they are most needed and will be used.)
- How to design safer surgical operations (**Chapter 7**).
- How to use safety checklists for making the operating room safer for patients and staff (**Appendix I**).
- Recommendations for waste management, a particularly difficult problem (**Chapter 8**).
- Guidelines for management of accidental exposure to HBV, HIV and HCV (**Chapter 7**).

In making these decisions, managers often must strike a balance between the importance of the problem and providing acceptable levels of safety for specific healthcare tasks. Two examples of situations frequently encountered by hospital managers in most developing countries are discussed in the following sections.

So-Called "Prophylactic Use" of Antibiotics

This issue warrants special consideration because it represents an inappropriate and costly misuse of valuable resources and also contributes to the growing problem of antibiotic resistance. For example, many service providers feel that because clients and patients have poor hygiene and/or

they are poorly nourished, giving a 5- to 7-day course of antibiotics—usually a tetracycline—will prevent infections following elective surgery. Not only have numerous articles documented that this does not work, but by definition this is not prophylactic antibiotic use.[1]

This is a management issue in which the education of professional staff (physicians and nurses) is extremely important and should include:

- Reviewing existing literature documenting that routine use of postoperative antibiotics in healthy patients undergoing elective surgery does not prevent infections (Ladipo et al 1991).
- Pointing out that the inappropriate use of antibiotics increases the prevalence of antibiotic resistance in the community and wastes precious resources.
- Reminding staff that when recommended infection prevention practices are conscientiously followed, routine postoperative antibiotics are not necessary (see **Chapter 7**).

Myths and Misconceptions about HIV/AIDS

The decisions and actions of healthcare staff are largely influenced by personal feelings, attitudes and beliefs, and their level of knowledge. For example, with the rapid emergence of the HIV/AIDS epidemic, especially in sub-Saharan Africa, parts of South Asia and the Caribbean, healthcare staff have become increasingly concerned about their own safety and about working in places where they come in contact with people who may be HIV-infected. This is a particularly difficult issue, especially when the risk to staff is associated with providing elective surgical procedures for health-related reasons, such as for family planning (e.g., voluntary sterilization, IUDs and Norplant implants), as opposed to medical-related services. These concerns can lead to either:

- adopting unnecessary and often expensive and excessive precautions; or
- taking unnecessary risks in the mistaken belief that for a given situation, there is little risk or that nothing can be done to minimize the risk (Flexner 1991; Klouda 1991).

Examples of unnecessary or excessive use of preventive practices include washing hands after shaking hands with people believed to be HIV-infected, and wearing examination gloves for any type of contact with patients known or believed to be HIV-infected. As a consequence, adequate supplies of valuable equipment (e.g., examination gloves) may not be available for situations where they are needed, such as for minor surgical procedures or vaginal exams for women in labor.

[1] Prophylactic antibiotic use is the provision of an antibiotic 30–60 minutes prior to starting a surgical procedure and ending not more than 6–12 hours postoperatively.

In some cases, however, health workers go to the other extreme and disregard proven protective practices, such as picking up suture needles with their gloved hand rather than with a forceps or recapping hypodermic needles. When recommended protective actions are not followed, healthcare workers place themselves, their patients and their fellow workers at risk, and the result is a management problem.

As mentioned above, preventing infections primarily involves education linked to behavior change interventions. Staff not only need to have correct information regarding risks and know how to avoid risks, but also they need to have appropriate risk-averting behavior demonstrated. In addition, personal concerns linked to the risk-taking behavior need to be addressed. For example, studies have shown that healthcare workers are often willing to change bad attitudes and work habits when they understand the reason for and importance of a new safety procedure (Raven and Haley 1982; Seto et al 1990). Unfortunately, it was also noted in these same studies that while positive behavioral changes may occur following training, such compliance often decreases again in a few days or weeks. Thus, in order to ensure continued compliance, management reinforcement is needed, as well as a monitoring system that ties results to overall performance indicators.

STAFF TRAINING

Initially, all levels of healthcare workers (e.g., nurses, physicians, housekeepers and cleaners) need to know why infection prevention is important. Key topics to be taught should include:

- The disease transmission cycle, routes of infection and how to break the cycle (see **Chapter 1** and **Figures 1-1** and **1-2**).
- Use of Standard Precautions when dealing with all patients, not just those who appear or are known to be infected (**Chapter 2**).
- Methods of minimizing disease transmission (i.e., hand hygiene, gloves and other PPE) as well as "hands-on" demonstrations covering, for example:

    - Handwashing and using a waterless, alcohol-based antiseptic handrub
    - Cleaning up a blood or body fluid spill
    - Giving an injection and disposing of sharps
    - Learning to suture with blunt-tipped needles

To have long-term effects, the initial training should be followed up, and monitoring should be targeted toward identifying and solving specific problems related to introducing the new process or procedure. General reminders regarding the importance of maintaining an infection-free environment for safer delivery of services also should be repeatedly emphasized.

MONITORING THE EFFECTIVENESS OF TRAINING

Regular monitoring of infection prevention practices and processes is important, not only to assess their effectiveness but also to determine the topics about which staff may need more training or review. To monitor effectiveness:

- Spot check how staff are performing any new procedures.
- Assess whether recommended practices are being followed.
- Note whether the necessary equipment and supplies are available and being used properly.

Based on the findings, future topics for training can be identified. **Table 19-1** is a sample checklist that managers can use to see whether recommended infection prevention practices are being followed.

Table 19-1. Checklist to Assess Whether Infection Prevention Guidelines Are Being Followed

Health facility: hospital: clinic: other: Date:

Type of health worker:: Evaluator:

(e.g., matron, sister, midwife, nursing assistant, etc.)

OBSERVATION	RESPONSE (Circle one) [N/A = Not applicable]		
OBSERVATION DURING FAMILY PLANNING PROCEDURES			
1. • High-level disinfected or examination gloves are worn for each vaginal examination	**YES**	**NO**	**N/A**
• Sterile (or high-level disinfected) gloves are used for voluntary sterilization or Norplant implants insertion	**YES**	**NO**	**N/A**
• High-level disinfected or examination gloves are worn for IUD insertion	**YES**	**NO**	**N/A**
2. Hands are thoroughly washed immediately:			
• Before putting on gloves	**YES**	**NO**	**N/A**
• After handling objects which might be contaminated	**YES**	**NO**	**N/A**
• After contact with blood or mucous membranes	**YES**	**NO**	**N/A**
• After removing gloves	**YES**	**NO**	**N/A**
3. Waste is disposed of by burning or burying	**YES**	**NO**	**N/A**
OBSERVATION OF SINGLE-USE NEEDLES, SCALPEL BLADES AND OTHER SHARP OBJECTS			
1. Needles, scalpel blades and other sharp objects are disposed of immediately after use	**YES**	**NO**	**N/A**
2. Needles, scalpel blades and other sharp objects are disposed of in a puncture-resistant container	**YES**	**NO**	**N/A**
Any other comments or observations?			
Any problems with implementation?			

OBSERVATION	RESPONSE (Circle one) [N/A = Not applicable]		
DECONTAMINATION AND CLEANING			
1. Waste items are disposed of according to guidelines	**YES**	**NO**	**N/A**
2. Blood spills are flooded with disinfectant and then wiped up	**YES**	**NO**	**N/A**
3. Instruments are decontaminated in a 0.5% chlorine solution immediately after use	**YES**	**NO**	**N/A**
4. Instruments are thoroughly cleaned and rinsed before sterilization or HLD	**YES**	**NO**	**N/A**
STERILIZATION			
5. What method of sterilization is used?			
• High-pressure steam (if **YES**, go to #6)	**YES**	**NO**	**N/A**
• Dry heat (if **YES**, go to #7)	**YES**	**NO**	**N/A**
6. When steam sterilizing, is the high-pressure steamer operating:			
• at 121°C (250°F)	**YES**	**NO**	**N/A**
• at a pressure of 106 kPa, 15 lb/in^2 (1 atmosphere)	**YES**	**NO**	**N/A**
• for at least 20 minutes for unwrapped items; 30 minutes for wrapped items	**YES**	**NO**	**N/A**
7. When using dry heat, are the instruments kept:			
• at 170°C (340°F) or 160°C (320°F) for sharps,	**YES**	**NO**	**N/A**
• at the required temperature (170°) for at least 1 hour, or	**YES**	**NO**	**N/A**
• at 160°C for 2 hours	**YES**	**NO**	**N/A**
HIGH-LEVEL DISINFECTION			
8. What method of high-level disinfection is used?			
• Boiling (if **YES**, go to #9)	**YES**	**NO**	**N/A**
• Steaming (if **YES**, go to #10)	**YES**	**NO**	**N/A**
• Chemical disinfectants (if **YES**, go to #11)	**YES**	**NO**	**N/A**
9. When boiling, are the instruments:			
• boiled for at least 20 minutes once boiling begins, and	**YES**	**NO**	**N/A**
• nothing is added after timing begins	**YES**	**NO**	**N/A**
10. When steaming, are the instruments:			
• steamed for at least 20 minutes once boiling begins, and	**YES**	**NO**	**N/A**
• nothing is added after timing begins	**YES**	**NO**	**N/A**
11. When chemical high-level disinfectants are used:			
• is an appropriate chemical used	**YES**	**NO**	**N/A**
• are items completely submerged	**YES**	**NO**	**N/A**
• are instruments soaked for at least 20 minutes	**YES**	**NO**	**N/A**
• are instruments rinsed with sterile/boiled water	**YES**	**NO**	**N/A**
Any other comments or observations?			
Any problems with implementation?			

MONITORING INFECTION PREVENTION PRACTICES

Keeping records of infections that occur in hospitals and clinics is a time-honored way of monitoring the effectiveness of infection prevention practices. In particular, keeping records on postoperative infections can help to identify breaks in recommended infection prevention practices. For example, when a series of similar infections occurs over a short time period, "trouble-shooting" should be done to identify the possible

cause(s). Assume a number of surgical wound infections occur in patients undergoing elective cesarean section. Trouble-shooting questions to consider include:

- Are recommended infection prevention practices being followed in the operating rooms? On the wards? (**Chapter 7**)
- Is the operative site (incision area) being cleaned preoperatively, especially if client hygiene is poor? (**Chapter 6**)
- Is an approved antiseptic at the correct concentration being used to prepare the operative site? (**Chapter 6**)
- Do any members of the surgical team have long fingernails? Wear colored nail polish? (**Chapter 3**)
- Are reused disposable surgical gloves being used? (**Chapter 14**)
- Are the infections linked to any particular surgical team? Or person? (**Chapters 3** and **7**)
- Are instruments and equipment being thoroughly cleaned prior to sterilization or high-level disinfection? (**Chapters 11** and **12**)
- Is the sterilizer (autoclave) working correctly? (**Chapter 11**)
- Is sterilization or high-level disinfection being timed correctly? (**Chapters 11** and **12**)

If the answer to any of these questions is "no," further information about the identified area(s) should be collected and the problem identified before deciding whether training, better equipment or management reinforcement is the corrective action needed. (This topic and the rationale and methods for investigating outbreaks are discussed further in **Chapter 28**.)

REFERENCES

Flexner C. 1991. Management of occupational exposures to HIV: An update. *AIDS Med Report* 4(2): 13–24.

Klouda A. 1991. Personal reference. International Planned Parenthood Federation. AIDS Prevention Unit: London.

Ladipo OA et al. 1991. Prevention of IUD-related pelvic infection: The efficacy of prophylactic doxycycline at IUD insertion. *Adv Contracept* 7(1): 43–54.

Lipscomb J and L Rosenstock. 1997. Health care workers: Protecting those who protect our health. *Infect Control Hosp Epidemiol* 18(6): 397–399.

Lynch P et al. 1997. *Infection Prevention with Limited Resources*. ETNA Communications: Chicago, pp 2–9.

Raven BH and RW Haley. 1982. Social influence and compliance of hospital nurses with infection control policies, in *Social Psychology and Behavioral Medicine*. Eiser RJ (ed). John Wiley & Sons, Inc.: New York, pp 413–438.

Seto WH et al. 1990. Brief report: The utilization of influencing tactics for the implementation of infection control policies. *Infect Control Hosp Epidemiol* 11(3): 144–150.

TWENTY

PREVENTING NOSOCOMIAL INFECTIONS

KEY CONCEPTS you will learn in this chapter include:

- What the most common types of nosocomial infections are
- What impact nosocomial infections have on healthcare
- How nosocomial infections increase the cost of healthcare
- Why preventing nosocomial infections is important

BACKGROUND

"Nosocomial infections are widespread. They are important contributors to morbidity and mortality. They will become even more important as a public health problem with increasing economic and human impact because of:

- **Increasing numbers and crowding of people.**
- **More frequent impaired immunity (age, illness and treatments).**
- **New microorganisms.**
- **Increasing bacterial resistance to antibiotics." (Ducel 1995)**

Nosocomial (hospital-acquired) infections are an important focus of infection prevention in all countries, but in developing countries they are a major cause of preventable disease and death. The most important are:

- urinary tract infections, pneumonia and diarrhea;
- infections following surgery or invasive medical procedures; and
- maternal and newborn infections.

The organisms causing most nosocomial infections usually come from the patient's own body (endogenous flora). They also can come from contact with staff (cross-contamination), contaminated instruments and needles, and the environment (exogenous flora). Because patients are highly mobile and hospital stays are becoming shorter, patients often are discharged before the infection becomes apparent (are symptomatic). In fact, a large portion of nosocomial infections in hospitalized patients—and all from ambulatory care facilities—become apparent only after the patients are discharged. As a consequence, it is often difficult to determine whether the source of the organism causing the infection is endogenous or exogenous.

Rates of nosocomial infections are markedly higher in many developing countries, especially for infections that are largely preventable (e.g., those following surgical procedures such as cesarean section). In these countries, nosocomial infection rates are high because of a lack of supervision, poor infection prevention practices, inappropriate use of limited resources and overcrowding of hospitals. Key contributing factors are:

- inadequate standards and practices for operating blood transfusion services (**Chapter 18**);
- increasing use of invasive medical devices (e.g., mechanical ventilators, urinary catheters and central intravenous lines) without proper training or laboratory support (**Chapters 17, 24** and **27**);
- use of contaminated intravenous fluids, especially in hospitals making their own IV solutions (**Chapter 24**);
- antibiotic resistance due to overuse of broad spectrum antibiotics; and
- unsafe and frequently unnecessary injections (**Chapters 7** and **24**).

The latter is most important. For example, after reviewing a number of studies, Simonsen et al (1999) concluded that more than 50% of injections in developing countries are unsafe (i.e., the needle, syringe or both are reused) and many injections are unnecessary (e.g., routine injections of vitamin B-12 or antibiotics). A major consequence of this is that an estimated 80,000 to 160,000 new HIV infections occur annually in sub-Saharan Africa, and even more cases of HBV and HCV occur worldwide each year as a result of unsafe injections (Kane et al 1999).

Understanding Nosocomial Infections

The role of Transmission-Based Precautions in minimizing the risk of nosocomial infections is detailed in **Chapter 21**. In subsequent chapters, information is presented regarding the epidemiology, microbiology, risk factors and practical measures for preventing nosocomial infections involving the urinary, gastrointestinal and respiratory systems (**Chapters 22, 26** and **27**) as well those following surgery (**Chapter 23**), the use of intravascular devices (**Chapter 24**) and maternal and newborn infections (**Chapter 25**). Also included is information on how to:

- manage food and water sources in hospitals and clinics in order to prevent food- and waterborne outbreaks; and
- assure a continuous source of clean and safe water for drinking and medical use (e.g., handwashing and instrument cleaning) (**Chapter 26**).

Finally, in **Chapter 28**, guidelines for monitoring (surveillance) of infection prevention practices and investigating outbreaks and exposures are briefly covered.

DEFINITIONS

- **Contaminated**. State of having been actually or potentially in contact with microorganisms. As used in healthcare, the term generally refers to the presence of microorganisms that could be capable of producing disease or infection.
- **Laboratory-acquired infection**. Nosocomial infection resulting from performance of laboratory activities by staff, regardless of how it occurred.
- **Nosocomial** or **hospital-acquired infection (terms used interchangeably)**. Infection that is neither present nor incubating at the time the patient came to the hospital. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.)
- **Occupational injury** or **infection**. Injury or infection acquired by healthcare staff while performing their normal duties.

FREQUENCY AND TYPE OF NOSOCOMIAL INFECTIONS

Nosocomial infections are a significant problem throughout the world and are increasing (Alvarado 2000). For example, nosocomial infection rates range from as low as 1% in a few countries in Europe and the Americas to more then 40% in parts of Asia, Latin America and sub-Saharan Africa (Lynch et al 1997). In 1987, a prevalence survey involving 55 hospitals in 14 developing countries in four WHO Regions (Europe, Eastern Mediterranean, South-East Asia and Western Pacific) found an average of 8.7% of all hospital patients had nosocomial infections. Thus at any time, over 1.4 million patients worldwide will have infectious complications acquired in the hospital (Tikhomirov 1987). In this survey the highest frequencies were reported from hospitals in the Eastern Mediterranean and South-East Asia Regions, 11.8% and 10% respectively (Mayon-White et al 1988). These rates most likely do not reflect the current situation because at that time the HIV/AIDS pandemic was just beginning. Moreover, the survey did not include any countries in Africa where nosocomial infection rates are much higher. They do, however, provide some guidance as to which types of nosocomial infections occur most frequently in developing countries. Surgical site infections, urinary tract infections and lower respiratory (pneumonia) infections were the leading types reported. This sequence differs somewhat from what is reported in the US, for example, where urinary and respiratory tract infections are the most common followed by surgical site infections (Emori and Gaynes 1993).

The WHO study and others also found that the highest prevalence of nosocomial infections occurs in intensive care units and acute care surgical and orthopedic wards. Not surprisingly, infection rates are higher among patients with increased susceptibility because of old age and the severity of the underlying disease. To this list should now be added those

hospitalized patients with decreased immunity due to AIDS and/or multidrug-resistant tuberculosis.

IMPACT OF NOSOCOMIAL INFECTIONS

Nosocomial infections add to functional disability, emotional stress and may, in some cases, lead to disabling conditions that reduce the quality of life. In addition, nosocomial infections have now become one of the leading causes of death (Ponce-de-Leon 1991). The impact of nosocomial infections takes on even more significance in resource-poor countries, especially those affected most by HIV/AIDS, because recent findings strongly suggest that unsafe medical care may be an important factor in transmitting HIV (Gisselquist et al 2002).

During the past 10–20 years little progress has been made in addressing the basic problems responsible for the increasing rates of nosocomial infections in many countries, and in some countries, conditions are actually worsening. Nosocomial infections increase the cost of healthcare in the countries least able to afford them through increased:

- length of hospitalization;
- treatment with expensive medications (e.g., antiretroviral drugs for HIV/AIDS and antibiotics); and
- use of other services (e.g., laboratory tests, X-rays and transfusions).

As a consequence, in resource poor countries, efforts to prevent nosocomial infections must assume even greater importance if progress is to be made in improving the quality of patient care in hospitals and other healthcare facilities.

PREVENTING NOSOCOMIAL INFECTIONS

Most of these infections can be prevented with readily available, relatively inexpensive strategies by:

- adhering to recommended infection prevention practices, especially hand hygiene and wearing gloves;
- paying attention to well-established processes for decontamination and cleaning of soiled instruments and other items, followed by either sterilization or high-level disinfection; and
- improving safety in operating rooms and other high-risk areas where the most serious and frequent injuries and exposures to infectious agents occur.

Unfortunately, not all nosocomial infections are preventable. For example, some reflect the influence of advanced age, chronic diseases such as uncontrolled diabetes, end-stage kidney disease or advanced pulmonary emphysema, severe malnutrition, treatment with certain drugs (e.g.,

antimicrobials, corticosteroids and other agents that decrease immunity), the increasing impact of AIDS (e.g., opportunistic infections) and irradiation.

REFERENCES

Alvarado CJ. 2000. *The Science of Hand Hygiene: A Self-Study Monograph*. University of Wisconsin Medical School and Sci-Health Communications. March.

Ducel G. 1995. Les nouveaux risques infectieux. *Futuribles*: 203: 5–32.

Emori TG and RP Gaynes. 1993. An overview of nosocomial infections, including the role of the clinical laboratory. *Clin Microbiol Rev* 6(4): 428–442.

Gisselquist D et al 2002. HIV infections in sub-Saharan Africa not explained by sexual or vertical transmission. *Int J STD AIDS* 13(10): 657–666.

Lynch P et al. 1997. *Infection Prevention with Limited Resources*. ETNA Communications: Chicago.

Kane A et al. 1999. Transmission of hepatitis B, hepatitis C and human immunodeficiency viruses through unsafe injections in the developing world: Model-based regional estimates. *Bull World Health Organ* 77(10): 801–807.

Mayon-White RT et al. 1988. An international survey of the prevalence of hospital-acquired infection. *J Hosp Infect* 11(Suppl A): 43–48.

Ponce-de-Leon S. 1991. The needs of developing countries and the resources required. *J Hosp Infect* 18 (Suppl A): 376–381.

Simonsen L et al. 1999. Unsafe injections in the developing world and transmission of bloodborne pathogens: A review. *Bull World Health Organ* 77(10): 789–800.

Tikhomirov E. 1987. WHO programme for the control of hospital infections. *Chemiotherapia* 6(3): 148–151.

TWENTY-ONE

ISOLATION PRECAUTION GUIDELINES FOR HOSPITALS

KEY CONCEPTS you will learn in this chapter include:

- What the reasons for the Transmission-Based Precautions are
- What Transmission-Based Precautions are designed to do
- What preventive processes and practices are recommended for each route of infection transmission
- How to effectively use Transmission-Based Precautions

BACKGROUND

Although the spread of infectious diseases in hospitals has been recognized for many years, understanding how to prevent nosocomial (hospital-acquired) infections and implementing policies and practices that are successful have been more difficult. The transmission of nosocomial infections requires three elements: a **source** of infecting microorganisms, a **susceptible host** and a **mode of transmission**.

The **human source** of nosocomial infections may be patients, hospital personnel or, less often, visitors. These people may have infectious diseases, be in the incubation period (no symptoms), or may be chronic carriers. Other **sources** of infecting microorganisms are inanimate objects that become contaminated (e.g., examination tables or medical instruments) and the environment, including air and water.

Susceptible hosts are those patients, hospital personnel and, less often, visitors who may become infected. Resistance among people to infecting microorganisms varies; for example, some are immune, others get infected and become asymptomatic carriers; and still others get infected and develop a clinical disease. Factors such as age, underlying diseases, treatment with certain drugs (e.g., antimicrobials, corticosteroids and other agents that decrease immunity) and irradiation play a role in this process.

The three main routes of infection transmission in hospitals are **airborne**, **droplet** and **contact**. An infecting microorganism, however, can be transmitted by more than one route. For example, varicella (chicken pox) is transmitted both by the airborne and contact route at different stages of the disease.

In previous sections (**Chapters 1** and **2**), the rationale and fundamentals of the new hospital-based isolation precautions have been laid out. The purpose of this chapter is to further explain how Transmission-Based Precautions are used in the hospital to minimize the risk of clients,

patients, visitors and staff becoming infected (i.e., developing a nosocomial infection) while dealing with the healthcare system.

DEFINITIONS

- **Airborne transmission**. Transfer of particles 5 μm or less in size into the air, either as airborne droplets or dust particles containing the infectious microorganism; can be produced by coughing, sneezing, talking or procedures such as bronchoscopy or suctioning; can remain in the air for up to several hours; and can be spread widely within a room or over longer distances. Special air handling and ventilation are needed to prevent airborne transmission.
- **Cohorting**. Practice of placing patients with the same active infectious disease (e.g., chicken pox)—but no other infection—in the same room or ward.
- **Colonization**. Pathogenic (illness- or disease-causing) organisms are present in a person (i.e., they can be detected by cultures or other tests) but are not causing symptoms or clinical findings (i.e., no cellular changes or damage). Coming in contact with and acquiring new organisms, while increasing the risk of infection, usually does not lead to infection because the body's natural defense mechanism (the immune system) is able to tolerate and/or destroy them. Thus, when organisms are transmitted from one person to another, colonization rather than infection is generally the result. Colonized persons, however, can be a major source of transfer of pathogens to other persons (cross-contamination) especially if the organisms persist in the person (chronic carrier), such as with HIV, HBV and HCV.
- **Contact transmission**. Infectious agent (bacteria, virus or parasite) transmitted directly or indirectly from one infected or colonized person to a susceptible host (patient), often on the contaminated hands of a health worker.
- **Droplet transmission**. Contact of the mucous membranes of the nose, mouth or conjunctivae of the eye with infectious particles larger than 5 μm in size that can be produced by coughing, sneezing, talking or procedures such as bronchoscopy or suctioning. Droplet transmission requires close contact between the source and the susceptible person because particles remain airborne briefly and travel only about 1 meter (3 feet) or less.
- **Nosocomial** or **hospital-acquired infection (terms used interchangeably)**. Infection that is neither present nor incubating at the time the patient came to the hospital. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.)

TRANSMISSION-BASED PRECAUTIONS

Note: Protective isolation of immunocompromised patients, such as those with AIDS, is not an effective way to reduce the risk of cross-infection (Manangan et al 2001).

The isolation guidelines issued by CDC in 1996 involve a two-level approach: **Standard Precautions,** which apply to **all clients** and **patients** attending healthcare facilities, and **Transmission-Based Precautions**, which apply primarily to **hospitalized patients** (Garner and HICPAC 1996). As briefly presented in **Chapter 1**, this system replaces the cumbersome disease-specific isolation precautions with three sets of Transmission-Based Precautions (air, droplet or contact).

> In all situations, whether used alone or in combination, Transmission-Based Precautions must be used in conjunction with the Standard Precautions (Garner and HICPAC 1996).

Airborne Precautions

These precautions are designed to reduce the nosocomial transmission of particles 5 μm or less in size that can remain in the air for several hours and be widely dispersed (**Table 21-1**). Microorganisms spread wholly or partly by the airborne route include tuberculosis (TB), chicken pox (varicella virus) and measles (rubeola virus). Airborne precautions are recommended for patients with either **known** or **suspected** infections with these agents. For example, an HIV-infected person with a cough, night sweats or fever, and clinical or X-ray findings in the lungs should go on airborne precautions until TB is ruled out.

Table 21-1. Airborne Precautions

Used in addition to Standard Precautions for a patient known or suspected to be infected with microorganisms transmitted by the airborne route.

PATIENT PLACEMENT

- Private room.
- Door closed.
- Room air is exhausted to the outside (negative air pressure) using fan or other filtration system.
- If private room not available, place patient in room with patient having active infection with the same disease, but with no other infection (cohorting).
- Check all visitors for susceptibility before allowing them to visit.

RESPIRATORY PROTECTION

- Wear surgical mask.
- If TB known or suspected, wear particulate respirator (if available).
- If chicken pox or measles:
  - Immune persons—no mask required.
  - Susceptible persons—do not enter room.
- Remove mask after leaving the room and place in a plastic bag or waste container with tight-fitting lid.

PATIENT TRANSPORT

- Limit transport of patient to essential purposes only.
- During transport, patient must wear surgical mask.
- Notify area receiving patient.

Adapted from: ETNA Communications 2000.

Where TB is prevalent, it is important to have a mechanism to quickly assess (triage) patients with suspected TB because delayed diagnosis, resulting in lack of isolation, has been shown to be an important factor in hospital-based transmission to other patients. In this situation, airborne precautions are the last defense in reducing the risk of TB transmission.

Droplet Precautions

These precautions reduce the risks for nosocomial transmission of pathogens spread wholly or partly by droplets larger than 5 μm in size (e.g., *H. influenzae* and *N. meningitides* meningitis; *M. pneumoniae*, flu, mumps and rubella viruses). Other conditions include diphtheria, pertussis (whooping cough), pneumonic plague and strep pharyngitis (scarlet fever in infants and young children).

Droplet precautions are simpler than airborne precautions because the particles remain in the air only for a short time and travel only a few feet; therefore, contact with the source must be close for a susceptible host to become infected (**Table 21-2**).

Table 21-2. Droplet Precautions

Use in addition to Standard Precautions for a patient known or suspected to be infected with microorganisms transmitted by large-particle droplets (larger than 5 μm).

PATIENT PLACEMENT

- Private room; door may be left open.
- If private room not available, place patient in room with patient having active infection with the same disease, but with no other infection (cohorting).
- If neither option is available, maintain separation of at least 1 meter (3 feet) between patients.

RESPIRATORY PROTECTION

- Wear mask if within 1 meter (3 feet) of patient.

PATIENT TRANSPORT

- Limit transport of patient to essential purposes only.
- During transport, patient must wear surgical mask.
- Notify area receiving patient.

Adapted from: ETNA Communications 2000.

Contact Precautions

These precautions reduce the risk of transmission of organisms from an infected or colonized patient through direct or indirect contact (**Table 21-3**). They are indicated for patients infected or colonized with enteric pathogens (hepatitis A or echo viruses), herpes simplex and hemorrhagic fever viruses and multidrug (antibiotic)-resistant bacteria. Interestingly, chicken pox is spread both by the airborne and contact routes at different stages of the illness. Among infants there are a number of viruses

transmitted by direct contact. In addition, Contact Precautions should be implemented for patients with wet or draining infections that may be contagious (e.g., draining abscesses, herpes zoster, impetigo, conjunctivitis, scabies, lice and wound infections).

Table 21-3. Contact Precautions

Use in addition to Standard Precautions for a patient known or suspected to be infected or colonized with microorganisms transmitted by direct contact with the patient or indirect contact with environmental surfaces or patient care items.

PATIENT PLACEMENT

- Private room; door may be left open.
- If private room not available, place patient in room with patient having active infection with the same microorganism, but with no other infection (cohorting).

GLOVING

- Wear clean, nonsterile examination gloves (or reprocessed surgical gloves) when entering room.
- Change gloves after contact with infectious material (e.g., feces or wound drainage).
- Remove gloves before leaving patient room.

HANDWASHING

- Wash hands with antibacterial agent, or use a waterless, alcohol-based antiseptic handrub, after removing gloves.
- Do not touch potentially contaminated surfaces or items before leaving the room.

GOWNS AND PROTECTIVE APPAREL

- Wear clean, nonsterile gown when entering patient room if patient contact is anticipated or patient is incontinent, has diarrhea, an ileostomy, colostomy or wound drainage not contained by a dressing.
- Remove gown before leaving room. Do not allow clothing to touch potentially contaminated surfaces or items before leaving the room.

PATIENT TRANSPORT

- Limit transport of patient to essential purposes only.
- During transport, ensure precautions are maintained to minimize risk of transmission of organisms.

PATIENT CARE EQUIPMENT

- Reserve noncritical patient care equipment for use with a single patient if possible.
- Clean and disinfect any equipment shared among infected and noninfected patients after each use.

Adapted from: ETNA Communications 2000.

Empiric Use of Transmission-Based Precautions

If there is any question of an infectious process in a patient without a known diagnosis, implementing Transmission-Based Precautions should be considered based on the patient's signs and symptoms (empiric basis) until a definitive diagnosis is made. Moreover, where healthcare resources, including laboratory testing, are limited, diagnosis-based isolation precautions are not helpful in practice. In these circumstances, the isolation system needs to be completely based on the clinical findings (signs and symptoms).

Examples of the "empiric use" of these precautions are illustrated in **Table 21-4**.

Table 21-4. Empiric Use of Transmission-Based Precautions (by signs and symptoms)

AIRBORNE	DROPLET	CONTACT
• Cough, fever and upper lobe chest findings (dullness and decreased breath sounds) • Cough, fever and chest findings in any area in HIV-infected person or at high risk for HIV • Rashes (vesicule or pustule)	• Severe, persistent cough during periods when pertussis is present in community • Meningitis (fever, vomiting and stiff neck) • Hemorrhagic rash with fever • Generalized rash of unknown cause	• Acute diarrhea in an incontinent or diapered patient • Diarrhea in adult with history of recent antibiotic use • Bronchitis and croup in infants and young children • History of infection with multiresistant organisms (except TB) • Abscess or draining wound that cannot be covered

A complete listing of the clinical syndromes or conditions warranting the empiric use of Transmission-Based Precautions is presented in **Table 21-5**.

The use of these precautions, including their empiric use in selected circumstances, is designed to reduce the risk of airborne-, droplet- and contact-transmitted infections between hospitalized patients and healthcare staff. To assist health workers in correctly implementing the appropriate precautions, **Table 21-6** provides a summary of the types of isolation precautions and the illnesses for which each type of precaution is recommended. In addition, **Appendix I** provides a complete listing of the types and duration of the isolation precautions needed for the vast majority of infectious diseases.

Table 21-5. Clinical Syndromes or Conditions to Be Considered for "Empiric Use" of Transmission-Based Precautions

CLINICAL SYNDROME OR CONDITION[a]	POTENTIAL PATHOGENS[b]	EMPIRIC PRECAUTIONS
Diarrhea		
Acute diarrhea with a likely infectious cause in an incontinent or diapered patient	Enteric pathogens[c]	Contact
Diarrhea in an adult with a history of recent antibiotic use	*Clostridium difficile*	Contact
Meningitis	*Neisseria meningitidis*	Droplet
Rash or exanthems, generalized, etiology unknown		
Petechial/ecchymotic with fever	*Neisseria meningitidis*	Droplet
Vesicular	Varicella (chicken pox)	Airborne and Contact
Maculopapular with coryza and fever	Rubeola (measles)	Airborne
Respiratory infections		
Cough/fever/upper lobe pulmonary infiltrate in an HIV-negative patient or a patient at low risk for HIV infection	*Mycobacterium tuberculosis*	Airborne
Cough/fever/pulmonary infiltrate in any lung location in an HIV-infected patient or a patient at high risk for HIV infection	*Mycobacterium tuberculosis*	Airborne
Paroxysmal or severe persistent cough during periods of pertussis activity	*Bordetella pertussis*	Droplet
Respiratory infections, particularly bronchiolitis and croup, in infants and young children	Respiratory syncytial or parainfluenza virus	Contact
Risk of multidrug-resistant microorganisms		
History of infection or colonization with multidrug-resistant organisms[d]	Resistant bacteria[d]	Contact
Skin, wound or urinary tract infection in a patient with a recent hospital or nursing home stay in a facility where multidrug-resistant organisms are prevalent	Resistant bacteria[d]	Contact
Skin or wound infection	*Staphylococcus aureus,* group A streptococcus	Contact

[a] Patients with the syndromes or conditions listed below may present with atypical signs or symptoms (e.g., pertussis in neonates and adults may not have paroxysmal or severe cough). The clinician's index of suspicion should be guided by the prevalence of specific conditions in the community, as well as clinical judgment.

[b] The organisms listed under the column "Potential Pathogens" are not intended to represent the complete, or even most likely, diagnoses, but rather possible etiologic agents that require additional precautions beyond Standard Precautions until they can be ruled out.

[c] These pathogens include enterohemorrhagic *Escherichia coli* O157:H7, *Shigella,* hepatitis A and rotavirus.

[d] Resistant bacteria judged by the infection control program, based on current state, regional or national recommendations, to be of special clinical or epidemiological significance.

Adapted from: Garner and HICPAC 1996.

Table 21-6. Summary of Types of Precautions and Patients Requiring the Precautions

Standard Precautions

Use Standard Precautions for the care of all patients.

Airborne Precautions

In addition to Standard Precautions, use Airborne Precautions for patients known or suspected to have serious illnesses transmitted by airborne droplet nuclei. Examples of such illnesses include:

- Measles
- Varicella (including disseminated zoster)[a]
- Tuberculosis[b]

Droplet Precautions

In addition to Standard Precautions, use Droplet Precautions for patients known or suspected to have serious illnesses transmitted by large particle droplets. Examples of such illnesses include:

Invasive *Haemophilus influenzae* type b disease, including meningitis, pneumonia, epiglottitis and sepsis

Invasive *Neisseria meningitidis* disease, including meningitis, pneumonia and sepsis

Other serious bacterial respiratory infections spread by droplet transmission, including:

- Diphtheria (pharyngeal)
- Mycoplasma pneumonia
- Pertussis
- Pneumonic plague
- Streptococcal (group A) pharyngitis, pneumonia, or scarlet fever in infants and young children

Serious viral infections spread by droplet transmission, including:

- Adenovirus[a]
- Influenza
- Mumps
- Parvovirus B19
- Rubella

Contact Precautions

In addition to Standard Precautions, use Contact Precautions for patients known or suspected to have serious illnesses easily transmitted by direct patient contact or by contact with items in the patient's environment. Examples of such illnesses include:

Gastrointestinal, respiratory, skin or wound infections or colonization with multidrug-resistant bacteria judged by the infection control program, based on current state, regional or national recommendations, to be of special clinical and epidemiologic significance.

Enteric infections with a low infectious dose or prolonged environmental survival, including:

- Clostridium difficile
- For diapered or incontinent patients: enterohemorrhagic Escherichia coli O157:H7, Shigella, hepatitis A or rotavirus

Respiratory syncytial virus, parainfluenza virus or enteroviral infections in infants and young children

Skin infections that are highly contagious or that may occur on dry skin, including:

- Diphtheria (cutaneous)
- Herpes simplex virus (neonatal or mucocutaneous)
- Impetigo
- Major (noncontained) abscesses, cellulitis or decubiti
- Pediculosis
- Scabies
- Staphylococcal furunculosis in infants and young children
- Zoster (disseminated or in the immunocompromised host)[a]

Viral/hemorrhagic conjunctivitis

Viral hemorrhagic infections (Ebola, Lassa, or Marburg)*

* See **Appendix I** for a complete listing of infections requiring precautions, including appropriate footnotes.

[a] Certain infections require more than one type of precaution.

[b] See CDC "Guidelines for Preventing the Transmission of Tuberculosis in Health-Care Facilities."

Adapted from: Garner and HICPAC 1996.

REFERENCES

ETNA Communications. 2000. Infection Control Signs, Available on: www.etnacomm.com

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80 and *Am J Infect Control* 24(1): 24–52.

Manangan LP et al. 2001. Infection control dogma: Top 10 suspects. *Infect Control Hosp Epidemiol* 22(4): 243–247.

TWENTY-TWO

PREVENTING URINARY TRACT INFECTIONS

KEY CONCEPTS you will learn in this chapter include:

- Why urinary tract infections are the most common type of nosocomial infections
- Why catheterization of the urinary system frequently leads to infection
- How to perform insertion, removal and replacement of an indwelling catheter
- How to minimize the risk of infection with an indwelling catheter

BACKGROUND

Urinary tract infections (UTIs) are the most common type of nosocomial (hospital-acquired) infections, accounting for 40% of all infections in hospitals per year (Burke and Zavasky 1999). In addition, several studies have reported that about 80% of nosocomial UTIs occur following instrumentation, primarily catheterization (Asher, Oliver and Fry 1986). Because nearly 10% of all hospitalized patients are catheterized, preventing UTIs is a major factor in decreasing nosocomial infections.

Organisms attacking any portion of the urinary system cause urinary tract infections: the kidneys (pyelonephritis), bladder (cystitis), prostate (prostatitis), urethra (urethritis) or urine (bacteriuria). Once bacteria infect any site, all other areas are at risk. The diagnosis of lower UTIs (cystitis and urethritis) is usually made on the basis of signs and symptoms and then confirmed by culture. Most episodes of short-term catheter-associated bacteriuria (greater than 10^5 organisms per mL of urine), however, are without symptoms. If present, symptoms usually consist of slight fever, burning, urgency and pain on urination. Similar symptoms or findings may occur in long-term catheterized patients, but these patients may also experience obstruction, urinary tract stones, renal failure and (rarely) bladder cancer (Warren 2000).

In upper UTIs (pyelonephritis), flank pain, fever, blood in the urine (hematuria) and other physical findings may be present. In frail, elderly patients, however, the typical signs and symptoms of a UTI may be absent. Moreover, bacteriuria, whether from an upper or lower UTI, is the most common cause of nosocomial gram-negative sepsis and has been linked to increased mortality (Platt et al 1982).

EPIDEMIOLOGY AND MICROBIOLOGY

In several prospective studies, rates of catheter-associated UTIs ranging from 9% to 23% have been reported (Johnson et al 1990). The wide range of rates may stem, in part, from recent improvements in care and technology (closed collection systems and better preventive care), as the highest rates were observed in studies prior to 1980. There is a greater risk of UTI with increased duration of catheterization. For example, about 50% of patients catheterized longer than 7–10 days typically develop an infection, but this increases to over 90% in patients catheterized more than 30 days (Garibaldi et al 1980). Moreover, if urine is allowed to drain into an open collection bag or container, **all** patients will develop bacteriuria within 4 days (with or without symptoms). Thus, the incidence of nosocomial UTIs depends, to a large extent, on the duration of catheterization and the type of drainage system (closed versus open).

Microbiology

Most nosocomial UTIs are caused by gram-negative coliform bacteria, particularly *Escherichia coli*, pseudomonas species, and organisms from the enterobacter group. Collectively they account for more than 80% of culture-positive UTIs (Haley et al 1985). While the most common organism is *E. coli*, infections with fungi, such as the candida species, have increased with the advent of HIV/AIDS and widespread use of broad spectrum antibiotics.

RISK FACTORS

Risk factors for nosocomial UTIs associated with catheterization can be broken down into those that are not alterable and those that are. Factors that are not alterable include: female gender, postpartum status, older age, severe underlying illness and high blood creatinine level. Factors that can be altered to reduce the risk of infection include: the wrong reason for catheterization, contamination during insertion, errors in catheter care and use of antibiotics.

Factors that can lead to bacteriuria and UTIs include:

- passage of organisms from the urine bag to the bladder (retrograde contamination) that occurs in 15–20% of patients with indwelling catheters (i.e., those left in place for several days or weeks); and
- ability of some organisms to grow on the outside or inside of the tubing and even in the urine itself.

Although these factors may not be alterable, preventing contamination of the collection bag, the bladder-to-bag tubing, the emptying tube on the bag or the mucosa lining the urethra can minimize the risk of infection.

REDUCING THE RISK OF NOSOCOMIAL URINARY TRACT INFECTIONS

Except for the end of the urethra or penis, the urinary system is normally sterile. The ability to completely empty the bladder is one of the most important ways the body has to keep the urine sterile and prevent UTIs. If the bladder empties completely during the voiding process, bacteria do not have the chance to infect tissue or grow and multiply in the bladder. Therefore, the normal defenses against a UTI are an unobstructed urethra, the voiding process and normal bladder mucosa. The insertion of a catheter, however, bypasses these defenses, introduces microorganisms from the end of the urethra or penis, and provides a pathway for organisms to reach the bladder.

Organisms may reach the bladder in two ways: through the inside of a catheter (i.e., the backward flow of urine) or by traveling up the space between the outer surface of the catheter and the urethral mucosa. Therefore, once the catheter is inserted, any back-and-forth movement of the catheter (e.g., raising the collection bag above the level of the bladder), or allowing urine to be collected in an open drainage system (bag or container) should be avoided because each of these activities potentially enables organisms to enter the bladder. The first way (backward flow of urine in the catheter) is the more common infection in men. The second (organisms migrating into the bladder along the outside of the catheter) is more common in women in part because of their shorter urethra. As a consequence, women are more likely to develop a UTI from organisms located in the vagina (Garibaldi et al 1980).

Placement of an indwelling catheter should be performed only when other methods of emptying the bladder are not effective, and it is particularly important to limit the duration as much as possible. The accepted indications for catheterization are:

- For short-term (days) management of incontinence (the inability to control urination) or retention (the inability to pass urine) not helped by other methods
- To measure urine output over several days in critically ill patients
- To instill medications
- For treatment of urinary outlet obstruction (blockage of the tube leading from the bladder to the outside, the urethra)
- For postoperative management of surgical patients with impaired bladder function (the most common routine use)

Other methods for management of urinary tract problems include: intermittent catheterization using a reusable “red rubber” straight catheter, condom catheters for male patients, adult diaper pads, bladder retraining and the use of drugs to stimulate urination.

Note: Indwelling catheters should not be used for the long-term management of incontinence.

Loss of control (incontinence) or inability to void (retention) may be managed better by straight (in and out) catheterization several times daily rather than by putting in an indwelling catheter. In addition, some patients can be trained to catheterize themselves for long-term care and can clean and high-level disinfect their own catheter by steaming it in a rice cooker or boiling it in a pot.

Procedures for Insertion, Removal, and/or Replacement of Urinary Catheters

Before inserting a catheter, check to be sure that it is being inserted for the right reason. For example, if a catheter is being inserted because of urinary retention, ask the patient if s/he has voided, the time of voiding and measure the height of the bladder. Also, before removing a catheter, check to be sure the doctor's orders are correct to avoid an error.

Insertion Procedure

STEP 1: Make sure that all of the following items are available:

- Sterile indwelling urinary catheter with a closed continuous drainage system, or a high-level disinfected or sterile straight catheter and a clean urine collection container
- High-level disinfected or sterile syringe filled with boiled or sterile water for blowing up the balloon of an indwelling catheter
- Pair of sterile or high-level disinfected surgical gloves
- Antiseptic solution (2% chlorhexidene gluconate or 10% povidone-iodine)
- Sponge forceps with gauze squares (2 x 2) or large cotton applicators
- Single-use packet of lubricant
- Light source (flashlight or lamp) if needed
- Basin of clean warm water, soap, a face cloth and a clean dry towel[1]
- Plastic bag or leakproof, covered waste container for disposal of contaminated items

Note: If using povidone-iodine, allow it to dry about 2 minutes because it only releases free iodine, the active antiseptic agent, slowly (**Chapter 6**).

STEP 2: Prior to starting the procedure:

- Have **women** separate their labia and gently wash the urethral area and inner labia.
- Have **men** retract their foreskin and gently wash the head of the penis and foreskin.

Note: If the patient is unable to wash her/himself, then a pair of clean examination gloves will be needed.

STEP 3: Wash hands with soap and clean water and dry with a clean dry towel or air dry. (Alternatively, if hands are not visibly soiled, apply 5 mL,

[1] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

about 1 teaspoonful, of a waterless, alcohol-based antiseptic handrub to both hands and vigorously rub the hands and between the fingers until dry.)

STEP 4: Put sterile or high-level disinfected gloves on both hands.

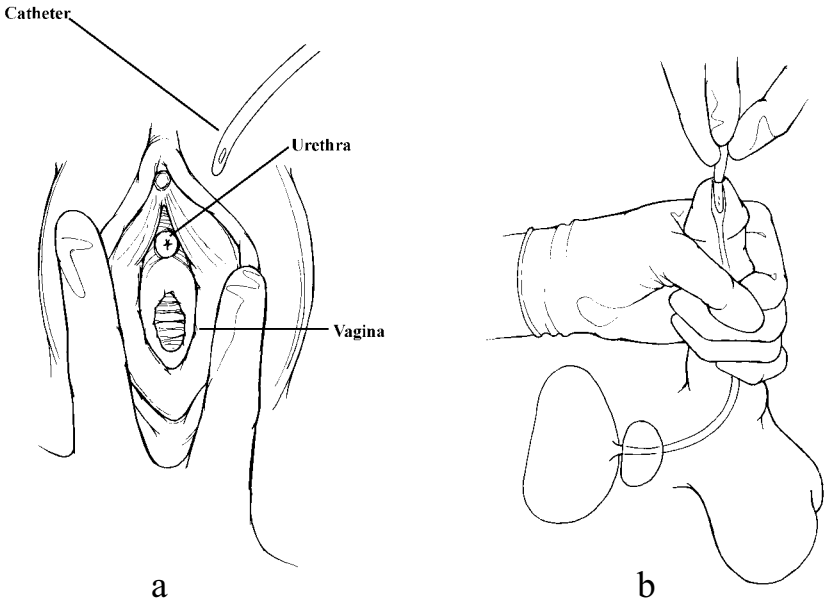
STEP 5: Use as small a catheter as consistent with good drainage.[2]

STEP 6: For health workers who are right-handed (dominant hand), stand on the patient's right side (and on the left side if left-handed).

STEP 7: **For women**, separate and hold the labia apart with the nondominant hand and prep the urethral area two times with an antiseptic solution using either cotton applicators or a sponge forceps with gauze squares (**Figure 22-1a**).

STEP 8: **For men**, push back the foreskin and hold the head of the penis with the nondominant hand; then prep the head of the penis and urethral opening two times with an antiseptic solution, using cotton applicators or a sponge forceps with gauze squares (**Figure 22-1b**).

Figure 22-1a and 1b. Catheterization Technique in Women and Men



Note: With indwelling catheters, do not disconnect the catheter from the drainage tube.

STEP 9: If inserting a **straight catheter**, grasp the catheter about 5 cm (2 inches) from the catheter tip with the dominant hand and place the other end in the urine collection container.

STEP 10: **For women**, gently insert the catheter as shown in **Figure 22-1a** about 5–8 cm (2–3 inches) or until urine flows. For children insert only about 3 cm (1.5 inches).

[2] No. 8–10 French is generally used for children and 14–16 for women. No. 16–18 is used for men unless a larger size is specified.

Note: Do not force catheter if resistance occurs.

STEP 11: For men, gently insert the catheter as shown in **Figure 22-1b** about 18–22 cm (7–9 inches) or until urine flows. For children insert only about 5–8 cm (2–3 inches).

STEP 12: If inserting an **indwelling catheter**, push another 5 cm (2 inches) after urine appears and connect the catheter to the urine collection tubing if not using a closed system.

STEP 13: **For an indwelling catheter**, inflate the balloon, pull out gently to feel resistance and secure the indwelling catheter properly to the thigh (for women) or lower abdomen (for men).

Note: If the catheter is accidentally inserted into the vagina, do not remove it. Reprep the urethral area with antiseptic solution and inset a new catheter into the urethra; then remove the one in the vagina.

STEP 14: **For straight (in and out) catheterization**, allow the urine to slowly drain into the collection container and then gently remove the catheter.

STEP 15: Place soiled items, including the straight catheter if it is to be disposed of, in a plastic bag or leakproof, covered waste container.

STEP 16: Alternatively, if a straight catheter is to be reused, place it in 0.5% chlorine solution and soak it for 10 minutes for decontamination.

STEP 17: Remove gloves by inverting and place them either in a plastic bag or waste container.

STEP 18: Wash hands or use an antiseptic handrub as above.

Removal and/or Replacement

STEP 1: Make sure all items are available (as in **Step 1** above if replacing an indwelling catheter):

- Pair of examination gloves (if replacing the catheter a pair of sterile or high-level disinfected gloves will be needed as well)
- Empty, high-level disinfected or sterile syringe for removing the fluid from the catheter balloon
- Sponge forceps with gauze squares (2 x 2) or large cotton applicators
- Plastic bag or leakproof, covered waste container for disposal of contaminated items

STEP 2: Have the patient wash the urethral area (women) or the head of the penis (men), or do it for them wearing a pair of clean examination gloves.

STEP 3: Wash hands or use an antiseptic handrub.

STEP 4: Put clean examination gloves on both hands.

STEP 5: With the empty syringe, remove the water from the catheter balloon.

STEP 6: **For women**, separate and hold the labia apart with the nondominant hand; then prep the urethral area two times with an antiseptic

solution using either cotton applicators or a sponge forceps with gauze squares and gently remove the catheter.

STEP 7: **For men**, push back the foreskin and hold the head of the penis with the nondominant hand; then prep the head of the penis and the area around the catheter two times with an antiseptic solution, using cotton applicators or sponge forceps with gauze squares and gently remove the catheter.

STEP 8: If you are just removing the catheter, then follow **Steps 15, 17** and **18** of the **Insertion Procedure**.

STEP 9: If you are replacing the indwelling catheter, follow **Steps 4 through 18** of the **Insertion Procedure**.

TIPS FOR PREVENTING INFECTIONS IN CATHETERIZED PATIENTS

- Remove the catheter as soon as possible.
- The catheter collection system should remain closed and not be opened unless absolutely necessary for diagnostic or therapeutic reasons.
- Caution the patient against pulling on the catheter.
- Urine flow through the catheter should be checked several times a day to ensure that the catheter is not blocked.
- Avoid raising the collection bag above the level of the bladder.
- If it becomes necessary to raise the bag above the level of the patient's bladder during transfer of the patient to a bed or stretcher, clamp the tubing.
- Before the patient stands up, drain all urine from the tubing into the bag.
- The urine drainage (collection) bags should be emptied aseptically; touching the tip of the emptying tube to the side of the collection bag or permitting the tip to touch the urine in the vessel should be avoided. Replace bags with new or clean containers when needed.
- If the drainage tubing becomes disconnected, do not touch the ends of the catheter or tubing. Wipe the ends of the catheter and tubing with an antiseptic solution before reconnecting them.
- Wash the head of the penis and urethral opening (men) or the tissue around the urethral opening (women) after a bowel movement or if the patient is incontinent.
- If frequent irrigation is required, the catheter should be changed.

Remember: Whenever a patient has a indwelling catheter in place, infection, including gram-negative septicemia, can occur, so check for signs of infection—back or flank pain, cloudy urine or fever.

What Does Not Work

Note: There is no evidence that daily perineal care (soap and water washing) reduces the risk of catheter-associated UTIs (Manangan et al 2001).

- Continuous irrigation of the bladder with antibiotics does not prevent bacteriuria and is associated with increased risk of resistant organisms (Warren et al 1978).
- While providing systemic antibiotics for brief periods (less than 5 days) may reduce the frequency of bacteriuria, it is not clear if it is worth the risk of drug reactions and the increased risk of resistant organisms (Burke, Larsen and Stevens 1986).
- Applying antiseptics (e.g., an iodophor such as Betadine®) or topical antibiotics to the perineal area (the urethral area for women and the head of the penis in men) does not reduce the risk of catheter-associated UTIs.

REUSING DISPOSABLE CATHETER MATERIALS

Note: After decontamination and cleaning, the catheter (straight and indwelling) should be carefully checked for cracks or tears and to be sure the balloon is not leaking.

In situations where resources are limited, the reuse of disposable straight and indwelling catheters and drainage tubing is acceptable if the recommended infection prevention practices are followed for decontamination, cleaning and high-level disinfection (i.e., by boiling or steaming) and air drying the devices in a high-level disinfected container (see **Chapter 9**).[3]

Note: If chemical disinfectants are used, the catheters and the tubing must be thoroughly rinsed at least three times with sterile or boiled water and care must be taken while rinsing not to contaminate the items.

The use of chemical disinfectants (e.g., glutaraldehydes) is not recommended for high-level disinfection (HLD). Making sure that all the disinfectant has been removed is difficult and time-consuming.

Drainage (collection) bags should be decontaminated and thoroughly cleaned and air dried before reuse. HLD is not necessary as long as care is taken to be sure that urine does not flow into the collection tubing (i.e., keep the level of the bag lower than the bladder and clamp off the tubing when moving the patient).

REFERENCES

Asher EF, BG Oliver and DE Fry. 1986. Urinary tract infections in the surgical patient. *Am Surg* 54(7): 466–469.

Burke JP and D Zavasky. 1999. Nosocomial urinary tract infections, in *Hospital Epidemiology and Infection Control*, 2nd ed. Mayhall CG (ed). Lippincott, Williams and Wilkins: Philadelphia, pp 173–187.

Burke JP, RA Larsen and LE Stevens. 1986. Nosocomial bacteriuria—estimating the potential for prevention by closed sterile drainage systems. *Infect Control* 7(Suppl 2): 96–99.

[3] To speed up drying out the inside of catheters and collection tubing, allow them to drain thoroughly before placing in the storage container. To do this, put high-level disinfected gloves on both hands and then carefully remove the item from the steamer or boiler with high-level disinfected forceps. While holding one end of the catheter with a gloved hand, allow the other end to hang down and shake it gently. When doing this, be careful that the catheter or tubing does not touch anything.

Garibaldi RA et al. 1980. Meatal colonization and catheter-associated bacteriuria. *N Engl J Med* 303(6): 316–318.

Haley RW et al. 1985. The nationwide nosocomial infection rate: A new need for vital statistics in U.S. hospitals. *Am J Epidemiol* 121(2): 182–295.

Johnson JR et al. 1990. Prevention of catheter-associated urinary tract infections with a silver oxide-coated urinary catheter: Clinical and microbiological correlates. *J Infect Dis* 162(5): 1145–1150.

Manangan LP et al. 2001. Infection control dogma: top 10 suspects. *Infect Control Hosp Epidemiol* 22(4): 243–247.

Platt R et al. 1982. Mortality associated with nosocomial urinary tract infection. *N Engl J Med* 307(11): 637–642.

Warren JW. 2000. Nosocomial urinary tract infections, in *Principles and Practices of Infectious Diseases*, 5th ed. Mandell JE et al (eds). Churchill Livingstone, Inc.: Philadelphia, pp 328–339.

Warren JW et al. 1978. Antibiotic irrigation and catheter-associated urinary tract infections. *N Engl J Med* 299(11): 570–573.

TWENTY-THREE

PREVENTING SURGICAL SITE INFECTIONS

KEY CONCEPTS you will learn in this chapter include:

- What the factors that affect the risk of nosocomial surgical site infections are
- How to reduce the risk of nosocomial surgical site infections
- What the rationale for antibiotic prophylaxis is
- When the use of prophylactic antibiotics is indicated
- What the recommendations for prevention of bacterial endocarditis are

BACKGROUND

Before the work of Joseph Lister and others in the 1860s, surgical patients commonly developed postoperative fever followed by purulent drainage from their incisions, sepsis and often death. The introduction of the principles of antisepsis by Lister and the acceptance of Pasteur's germ theory in the late nineteenth century led to a marked decrease in wound infection rates. These discoveries also radically changed surgery from an activity associated with infection and death to one of preventing suffering and prolonging life. In the twentieth century, the two key factors that have enabled surgical advances, such as open heart surgery and kidney transplants, to become routinely possible and safe are improved anesthesia and scientifically sound infection prevention practices.

Despite improvements in operating room practices, instrument sterilization methods, better surgical technique and the best efforts of infection prevention practitioners, surgical site infections (SSIs) remain a major cause of nosocomial (hospital-acquired) infections—and rates are increasing globally (Alvarado 2000). Moreover, in countries where resources are limited, even basic life-saving operations, such as appendectomies and cesarean sections, are associated with high infection rates and mortality. In these countries, therefore, it makes sense to focus on preventing SSIs in those procedures most frequently performed and/or those having the highest SSI rates.

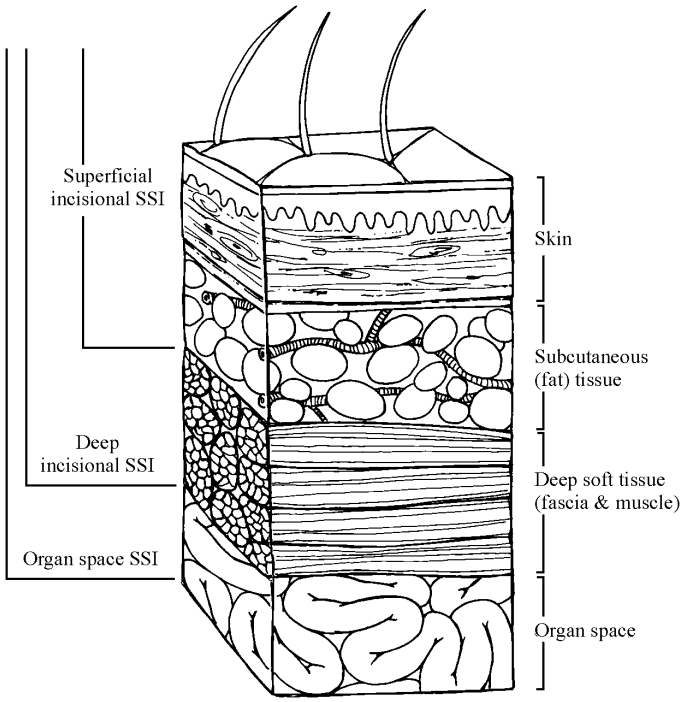
To reduce the risk of nosocomial SSIs in developing countries, a systematic but realistic approach must be applied with awareness that this risk is influenced by characteristics of the patient, the operation, the healthcare staff and the hospital. In theory, reducing risk is relatively simple and inexpensive, especially when compared to the cost of the infections themselves, but in practice it requires commitment at all levels of the healthcare system. And, as noted in **Chapter 20**, neither the basic

problems responsible for the high nosocomial rates (i.e., lack of training, supervision, infrastructure and resources) nor the recommended solutions have changed over the past 10–20 years in most developing countries.

DEFINITIONS

- **Organ/Space SSI**. Any part of the body other than the incised body wall parts that were opened or handled during an operation.
- **Surgical site infections (SSI)**. Either an **incisional** or **organ/space** infection occurring within 30 days after an operation or within 1 year if an implant is present. As shown in **Figure 23-1**, incisional SSIs are further divided into **superficial incisional** (only involves skin and subcutaneous tissue)[1] and **deep incisional** (involves deeper soft tissue, including fascia and muscle layers).[2]

Figure 23-1. Cross-Section of Abdominal Wall Showing CDC Classifications of Surgical Site Infection



Adapted from: Horan et al 1992.

[1] Does not include stitch abscess, infection of episiotomy or newborn circumcision, or infected burn wound. Specific criteria are used for identifying these infections and reporting them.

[2] For confirmation of all SSIs, clinical findings (signs or symptoms of infections) and/or laboratory test results (organism isolated from aseptically obtained culture) are required.

The surgical wound classification system includes four categories:

- **Class I—clean**. Uninfected operative wound with no inflammation and in which the respiratory, gastrointestinal (GI), genital and urinary tracts were not entered. Clean wounds are closed at surgery and, if necessary, drained with closed drainage.
- **Class II—clean-contaminated**. Wound in which the respiratory, GI, genital or urinary tract(s) were entered under controlled conditions but without unusual contamination or spillage of contents.
- **Class III—contaminated**. Open, fresh accidental wound or an operation with a major break(s) in aseptic technique (e.g., open cardiac massage) or gross spillage from the GI tract. Also included are incisions in which acute, nonpurulent inflammation is found.
- **Class IV—dirty or infected**. Old wounds with dead tissue and those that involve existing clinical infection or a perforated bowel, suggesting that the pathogens causing the postoperative infection were present in the wound before the surgery.

EPIDEMIOLGY AND MICROBIOLOGY

Among surgical patients, SSIs are the most common nosocomial infection, accounting for about a third of all such infections. In most studies about two thirds of these can be classified as superficial incisional, while the remaining involve either organs or spaces entered during surgery or are deep incisional SSIs. On average, having an SSI increases a patient's hospital stay by 7–10 days, with organ/space and deep incisional SSIs accounting for the longest stays and highest costs.

Organisms associated with SSIs vary with the type of procedure and the anatomic location of the operation. *Staphylcoccus aureus* (coagulase-negative staphylococci), enterococcus species and *Escherichia coli* are the three most frequently isolated pathogens. An increasing number of SSIs are caused by antimicrobial-resistant pathogens, and the incidence of fungal SSIs has risen significantly in the last decade in part because of the dramatic increase in the number of HIV/AIDS patients. For most SSIs, the source of the pathogen(s) comes from the patient's skin, mucous membranes or bowel and rarely from another infected site in the body (endogenous sources). Exogenous sources of SSI pathogens are occasionally responsible. These include:

- organisms from members of the surgical team (e.g., hands, nose or other body parts);
- contaminated surfaces in the operating room, even the air; and
- contaminated instruments, surgical gloves or other items used in the surgery.

Exogenous organisms are primarily aerobic staphylococci or streptococci species (with the exception of tetanus endospores). Although fungi are widely present in the environment, they rarely cause SSIs.

The mechanisms by which microorganisms infect tissue and produce disease are complex and incompletely understood. For example, some pathogens may contain or produce toxins and other substances that increase their ability to invade a patient's tissue, produce damage or survive in the tissue.

PATHOGENESIS

By the end of an operation, bacteria and other microorganisms contaminate all surgical wounds, but only a small number of patients actually develop a clinical infection (Fry 2003). Infection does not develop in most patients because their defense mechanisms effectively eliminate the contaminating organisms at the surgical site. Whether a potential infection occurs depends on several factors, with the most important being:

- number of bacteria entering the wound;
- type and virulence (ability to cause infection) of the bacteria;
- host defense mechanisms (e.g., effectiveness of inflammatory response and status of the immune system); and
- external factors, such as being in the hospital several days before surgery or the operation lasting more than 4 hours.

Two factors that can help minimize the number of organisms entering the wound are the skill and experience of the surgeon and use of good surgical technique. Both are important because if a surgical site is contaminated with more than 10^5 (100,000) organisms per gram of tissue, the risk of SSI is markedly increased (Krizek and Robson 1975). The dose required for infection can be even lower, however, if foreign material is present at the site (e.g., only 10^2 or about 100 staphylococci are enough if silk suture is used for closure or to control bleeding) (James and MacLeod 1961).

While the type and virulence of the bacteria cannot be controlled, the other factors can to a large extent. For example, tissue injury caused by making the wound incision triggers a chain of events, called the inflammatory response, that take place even before bacterial contamination occurs. The effectiveness of the inflammatory response to mobilize patient defense mechanisms (e.g., activation of various types of white blood cells that contain and destroy the bacteria before infection can occur) depends to large extent on the patient's general health, age, obesity, smoking, some chronic diseases and the status of the immune system.

RISK FACTORS

Table 23-1 lists the most widely accepted patient and operative characteristics that may increase the risk of an SSI. What is interesting about this list is how short it is. Of the many possible human conditions and surgical practices, it is surprising how few have been proven to independently influence the risk of infection. In part this is due to the complex nature of SSIs and to the great difficulty in designing and conducting studies that accurately isolate the effect of a single factor.

Table 23-1. Patient and Operation Characteristics That May Influence the Risk of Developing a Surgical Site Infection

PATIENT

Nutritional status, poor
Diabetes, uncontrolled
Smoking or use of other tobacco products
Obesity
Coexistent infections at a remote body site
Colonization with microorganisms
Altered immune response (HIV/AIDS and chronic corticosteroid use)
Length of preoperative stay

OPERATION

Preoperative shaving
Preoperative skin prep
Duration of operation
Antimicrobial prophylaxis
Operating room ventilation
Instrument processing (cleaning, HLD or sterilization)
Foreign material in the surgical site
Surgical drains
Surgical technique

- Poor hemostasis
- Failure to obliterate dead space
- Tissue trauma

Adapted from: SHEA, APIC, CDC and SIS 1990.

Patient Factors

- **Obesity** increases risk substantially when the subcutaneous abdominal fat layer exceeds 3 cm (1.5 inches) (Nyström et al 1987). The risk is increased by the need for a larger incision, decreased circulation to the fat tissue or the technical difficulty of operating through a large fat layer.
- **Infection at another site** may increase the risk of spreading infection through the bloodstream.
- **Immunocompromised patients** (e.g., those with HIV/AIDS, those with chronic corticosteroid use such as occurs with asthma and heavy

smokers or users of other tobacco products) are at significantly greater risk of SSIs.

- **Malnutrition** may or may not be a contributing factor. Unfortunately, most studies have not been conducted in developing countries where severe malnutrition is more common.
- **Age, race, socioeconomic status and chronic diseases**, such as diabetes and malignancy, are difficult to assess because they are frequently associated with other factors that independently contribute to risk. For example, age over 70 may be accompanied by decreased defense mechanisms, poor nutrition and anemia.

When possible, the effects of conditions that might complicate surgical recovery should be corrected or stabilized preoperatively. For example:

- Although diabetes and high blood pressure are not independent risk factors, they should be under control before elective surgery.
- Smoking or use of other tobacco products should be stopped at least 30 days before elective surgery if possible.
- Patients with infections remote to the surgical site should be treated if possible or their surgery postponed.
- Women using combined (estrogen- and progestogen-containing) contraceptives (oral or injectable) should be switched to a nonhormonal method at least 30 days before major elective surgery to minimize the risk of deep vein thrombophlebitis and nonfatal pulmonary embolism (Blumenthal and McIntosh 1996).

REDUCING THE RISK OF SURGICAL SITE INFECTIONS

In 1999, CDC issued guidelines for reducing the risk of SSIs based on existing scientific data, theoretical rationale and applicability. A copy of these recommendations, including the strength of the scientific information (Category I or II) on which they are based, is presented in **Appendix J**. Because these recommendations are intended to be used in US healthcare facilities, administrators and health professional staff in developing countries will need to carefully review, accept or modify them according to what is possible, practical and doable within their resource setting. While the vast majority of these recommendations are applicable and doable even in limited resource settings, some are not. For example, recommendations regarding Intraoperative Operating Room Ventilation (Section 2a) that require positive-pressure ventilation, provision of 15 air exchanges per hour and filtration of all air (fresh or recirculated)—all Category 1B recommendations—may not be financially possible. Other recommendations that may need to be modified, depending on available resources and the nature of the surgical procedure, include instrument sterilization recommendations (Section 2d) and the use of sterile surgical attire and drapes that are fluid-resistant (Section 2e).

In addition, some factors that may affect the risk of infection have either not been studied or the results of existing studies are inconclusive (e.g., members of the surgical team wearing nail polish). As a consequence, for these factors either no recommendation is provided in the guidelines or they are not dealt with at all. A few of the most notable omissions include whether or not to:

- limit traffic flow (i.e., the number of people in the operating room) during surgery;
- wear soiled surgical clothing from case to case;
- perform more than one operation in the same room, including the use of shared personnel;
- cover a clean incision closed at surgery beyond 48 hours; or
- advise the patient to bathe or shower after surgery without a dressing.

For most of these, standard practice would advise against doing them. With regard to care of the incision, it is generally believed that postoperative care has only minimal effect on the risk of SSIs. This belief is based on the assumption that wounds begin to heal immediately and after 48 hours do not to require a dressing or will not become infected by showering or bathing. This assumption, however, may not be valid, especially in limited-resource settings where hygiene is poor and the quality of tap water is questionable or frankly contaminated. For example, a 1991 report by Lowry et al documented that an outbreak of Legionnaire's disease was related to contaminated tap water used for washing around surgical wounds. Thus, where the likelihood of wound contamination is high and the quality of tap water poor, it is probably advisable to keep the incision clean, dry and covered. Bathing or showering should be avoided until the incision is nearly healed (5–7 days).

Note: Putting topical antibiotic ointments on closed skin incisions does not decrease the risk of SSIs. (Fry 2003).

Recommendations for postoperative care are quite different for a surgical incision that is either:

- left open at the skin level for a few days (usually 4–5 days) before it is closed (delayed primary closure); or
- left open to heal by secondary intention (i.e., healing from the base upwards until reaching the surface).

Note: Healthy tissue growth is damaged when the dry gauze is removed; therefore, moisten the dry gauze with sterile normal saline before removing it.

In both situations, the incision initially should be packed and covered with a sterile, moist gauze dressing and changed regularly.

- If gauze dressings moistened with sterile normal saline are used, the dressing should be changed using aseptic technique (sterile or high-level disinfected gloves) every 8 hours to prevent the gauze from drying out.

Remember: Wash hands, or use an antiseptic handrub, before putting on gloves and after taking them off to avoid exposure to blood and other potentially infected body fluids and to decrease the risk of cross-contamination.

- If sterile gauze filled with petroleum jelly or other moistening agents is used to pack and cover the incision, it can be changed less often (24–48 hours), depending on the type of wound and the manufacturer's directions.

Unless the dressing and surrounding area can be kept dry, the patient should not bathe or shower while the incision is packed and covered with a dressing (or at least until granulation tissue is present in a wound healing by secondary intention).

Other Factors

- **Prolonged preoperative hospitalization** exposes patients to hospital flora, including multidrug-resistant organisms. Completing presurgical evaluations and correcting underlying conditions **before admission** to a hospital decreases this risk. Also, performing elective surgery, where feasible, in ambulatory surgery centers rather than acute care hospitals decreases the risk of exposure to hospital flora.
- **Preoperative hair removal** should be avoided if it is unnecessary. If hair must be removed, clip it with scissors just before the surgery. Shaving is a proven risk factor for SSIs (Cruse and Foord 1980).
- **Wide prepping of the proposed incision site** with antiseptic solution preoperatively helps keep microorganisms from migrating into the wound (breakthrough) if the site towels or drapes become wet during surgery (**Chapter 5**).
- **Good surgical technique** minimizes tissue trauma, controls bleeding, eliminates dead space, removes dead tissue and foreign bodies, uses minimal suture and maintains adequate blood supply and oxygenation. Specifically, it is important to:
  - handle soft tissue gently to avoid crushing that can result in tissue death (necrosis);
  - use electrocautery sparingly to control bleeding because it leaves behind dead tissue that is more likely to become infected;
  - use absorbable suture whenever possible because permanent suture, especially silk suture, reduces the number of bacteria necessary to cause infection (James and MacLeod 1961); and
  - use closed suction drains that exit through a separate stab wound to help prevent accumulation of tissue fluid in the dependent portion of the wound. Preventing this is especially important in obese patients and may reduce SSIs (Fry 2003). (Passive drains, such a Penrose drain, exiting through the bottom of the incision should not be used.)
- **Increased length of surgical procedures** is associated with increased risk of SSIs. It is estimated that the infection rate nearly doubles with each hour of surgery (Cruse and Foord 1980.)
- **Prompt discharge postoperatively**, provided patients are able to return to homecare, reduces the risk of infection as well.

These factors, coupled with the experience and skill of the surgeon and assistant, are known to reduce the risk of SSIs.

ANTIBIOTIC PROPHYLAXIS IN SURGERY

The use of antibiotics preoperatively can reduce the rate of infection, particularly wound infections, after certain operations. The benefit, however, must be weighed against the risks of toxic and allergic reactions, the emergence of resistant bacteria, drug interactions, superinfection and cost (Nichols 2001). For example, it is estimated that 5% of patients receiving an antibiotic will have a serious reaction to the drug. In general, antibiotic prophylaxis is recommended only for procedures with high infection rates and those in which the consequences of infection are especially serious. The recommendations for when to consider prophylactic antibiotics in general surgical, gynecologic and obstetric patients are outlined in **Table 23-2**.

Guidelines for Choosing a Prophylactic Antibiotic

Ideally the prophylactic drug(s) should be directed against the most likely infecting organisms, but need not kill or inactivate all pathogens. For most procedures, an inexpensive, first- or second-generation cephalosporin, such as cefazolin (Ancef®), which has a moderately long half-life and is active against staphylococci and streptococci, has been effective when given intravenously (IV) 30 minutes before surgery. Exceptions are for an appendectomy, where cefoxitin (Mefoxin®) or cefotetan (Cefotan®) is preferred because they are more active than cefazolin against bowel anaerobic organisms.

Where methicillin-resistant staphylococci are important postoperative pathogens, vancomycin (Vancocin®) can be used, but routine use for prophylaxis should be avoided because it may promote the emergence of resistant organisms. Also, third- and fourth-generation cephalosporins (e.g., ceotaxime or cefepime) should not be used for routine surgical prophylaxis because:

- they are expensive, some are less active than cefazolin against staphylococci;
- their spectrum of activity includes organisms rarely encountered in elective surgery; and
- their widespread use may promote the emergence of resistance.

Table 23-2. Prevention of Wound Infection and Sepsis in Surgical Patients

NATURE OF OPERATION	LIKELY PATHOGENS	RECOMMENDED DRUGS	ADULT DOSAGE BEFORE SURGERY
Gastrointestinal			
Colorectal	Enteric gram-negative bacilli, anaerobes, enterococci	*Oral*: neomycin plus erythromycin base[1]	
		IV: cefoxitin or	1–2 grams IV
		cefotetan	1–2 grams IV
		OR cefazolin plus	1–2 grams IV
		metronidazole	0.5 grams IV
Appendectomy	Enteric gram-negative bacilli, anaerobes, enterococci	cefoxitin or	1–2 grams IV
		cefotetan	1–2 grams IV
Genitourinary	Enteric gram-negative bacilli, enterococci	*High risk[2] only*: ciprofloxacin	500 mg PO or 400 mg IV
Gynecologic and Obstetric			
Vaginal or abdominal hysterectomy	Enteric gram-negative, anaerobes, group B strep, enterococci	cefazolin or	1–2 grams IV
		cefotetan or	1–2 grams IV
		cefoxitin	1 gram IV
Cesarean section	same as for hysterectomy	*High risk[3] only*: cefazolin	1 gram IV after cord clamping
Abortion	same as for hysterectomy	*First trimester, high risk[4]*:	2 million units IV
		aqueous penicillin G	300 mg PO[5]
		OR doxycycline	1 gram IV
		Second trimester: cefazolin	
Contaminated Surgery[6]			
Ruptured viscus	Enteric gram-negative bacilli, anaerobes, enterococci	cefoxitin or	1–2 grams IV q6h
		cefotetan plus or minus	1–2 grams IV q12h
		gentamicin	1.5 mg/kg IV q8h
		OR clindamycin plus	600 mg IV q6h
		gentamicin	1.5 mg/kg IV q8h
Traumatic wound	*S. aureus*, group A strep, clostridia	cefazolin[7]	1–2 grams IV q8h

[1] After appropriate diet and enemas, one gram of each at 1 pm, 2 pm and 11 pm the day before an 8 am operation.
[2] Urine culture positive or unavailable, preoperative catheter, transrectal prostatic biopsy.
[3] Active labor or premature rupture of membranes.
[4] Patients with previous pelvic inflammatory disease, previous gonorrhea or multiple sex partners.
[5] Divided into 100 mg 1 hour before the abortion and 200 mg one half hour after.
[6] For contaminated or "dirty" surgery, therapy should usually be continued for about 5 days. Ruptured viscus in postoperative setting (dehiscence) requires antibacterials to include coverage of nosocomial pathogens.
[7] For bite wounds in which likely pathogens may also include oral anaerobic organisms, such as *Eikenella corrodens* (human) or *Pasteruella multocida* (dog and cat), use ampicillin/sulbactam (*Unsayn®*). This antibiotic can also be used for penetrating intracranial wounds, including gunshot injuries.

Adapted with special permission from: *The Medical Letter* 2001.

Number of Doses In most instances, a single intravenous (IV) dose of an antibiotic completed 30 minutes or less before the skin incision provides adequate tissue levels throughout the operation. (If vancomycin is used, at least 1 hour is required.) Clearly the concept of "on call" infusion of prophylactic antibiotics is not acceptable because delays in starting the operation can occur, resulting in ineffective tissue levels when the surgery actually does start. If surgery is prolonged (more than 4 hours), major blood loss occurs or an antibiotic with a short half-life such as cefoxitin is used, one or more additional doses should be given during the procedure.

PREVENTION OF BACTERIAL ENDOCARDITIS

The risk of endocarditis is considered high in patients with previous endocarditis, prosthetic heart valves, complex congenital heart disease such as tetralogy of Fallot or surgically constructed pulmonary shunts or tubing (conduits). Virdans streptococci are the most common cause of endocarditis after dental or upper respiratory procedures, while enterococci are most frequently found following GI or genitourinary procedures.

Although the effectiveness of antimicrobial prophylaxis in preventing endocarditis has never been established by controlled clinical trials in humans (Level 1 evidence), many physicians believe their use before procedures that may cause brief periods of bacteremia is protective. The drugs and dosages in **Table 23-3** are based on those recommended by the American Heart Association (Dajani et al 1997).

Table 23-3. Endocarditis Prophylaxis

	DOSAGE FOR ADULTS	DOSAGE FOR CHILDREN*
DENTAL AND UPPER RESPIRATORY PROCEDURES		
Oral		
Amoxicillin (Amoxi®)	2 grams 1 hour before procedure	50 mg/kg 1 hour before procedure
Penicillin allergy: Clindamycin (Cleocin®) OR Azithromycin (Zithromax®)	600 mg 1 hour before procedure 500 mg 1 hour before procedure	20 mg/kg 1 hour before procedure 15 mg/kg 1 hour before procedure
Parenteral (for patients unable to take oral drugs)		
Ampicillin (Omnipen®)	2 grams IM or IV within 30 minutes before procedure	50 mg/kg IM or IV within 30 minutes before procedure
Penicillin allergy: Clindamycin	600 mg IV within 30 minutes before procedure	20 mg/kg IV within 30 minutes before procedure
GASTROINTESTINAL AND GENITOURINARY PROCEDURES		
Oral		
Amoxicillin	2 grams 1 hour before procedure	50 mg/kg 1 hour before procedure
Parenteral		
Ampicillin[1]	2 grams IM or IV within 30 minutes before procedure	50 mg/kg IM or IV within 30 minutes before procedure
Penicillin allergy: Vancomycin (Vancocin®)	1 gram IV infused *slowly over 1 hour* beginning 1 hour before procedure	20 mg/kg IV infused *slowly over 1 hour* beginning 1 hour before procedure
± Gentamicin[2] (Garamycin®)	1.5 mg/kg (120 mg max.) IM or IV within 30 minutes before procedure	1.5 mg/kg IM or IV within 30 minutes before procedure

* Should not exceed adult dosage.
[1] High-risk patients given parenteral ampicillin before the procedure should receive a dose of ampicillin 1 gram IM or IV or a dose of amoxicillin 1 gram orally 6 hours afterwards.
[2] Gentamicin should be added for patients with a high risk of endocarditis.

Adapted with special permission from: *The Medical Letter* 2001, citing recommendations by Dajani et al 1997.

REFERENCES

Alvarado CJ. 2000. *The Science of Hand Hygiene: A Self-Study Monograph*. University of Wisconsin Medical School and Sci-Health Communications. March.

Blumenthal P and N McIntosh. 1996. Combined (estrogen and progestin) contraceptives, in *PocketGuide for Family Planning Service Providers*, 2nd ed. JHPIEGO Corporation: Baltimore, MD, pp 86–114.

Cruse PJE and R Foord. 1980. The epidemiology of wound infection: A 10 year prospective study of 62,939 wounds. *Surg Clin North Am* 60(1): 27–40.

Dajani AS et al. 1997. Prevention of bacterial endocarditis: Recommendations of the American Heart Association. *JAMA* 277(22): 1794–1801.

Fry DE. 2003. Surgical site infection: Pathogenesis and prevention. *Medscape* (February 19). Available at: www://medscape.com/viewprogram /2220_pnt.

Horan TC et al. 1992. CDC definitions of nosocomial surgical site infections, 1992: A modification of CDC definitions of surgical wound infections. *Infect Control Hosp Epidemiol* 13(10): 606–608.

James RC and CJ MacLeod. 1961. Induction of staphylococcal infections in mice with small inocula introduced on sutures. *Br J Exp Pathol* 42: 266–272.

Krizek TJ and MC Robson. 1975. Evolution of quantitative bacteriology in wound management. *Am J Surg* 130(5): 579–584.

Lowry PW et al. 1991. A cluster of legionella sternal-wound infections due to postoperative topical exposure to contaminated tap water. *N Engl J Med* 324(2): 109–113.

The Medical Letter. 2001. Antimicrobial prophylaxis in surgery. *The Medical Letter* 43: 1116–1117.

Nichols RL. 2001. Preventing surgical site infections: A surgeon's perspective. *Emerg Infect Dis* 7(2): 220–224.

Nyström P et al. 1987. Incisional infection after colorectal surgery in obese patients. *Acta Chir Scand* 153(3): 225–227.

SHEA, APIC, CDC and SIS. 1990. Consensus paper on the surveillance of surgical wound infections. *Infect Control Hosp Epidemiol* 18(5): 599–605.

TWENTY-FOUR

PREVENTING INFECTIONS RELATED TO USE OF INTRAVASCULAR DEVICES

KEY CONCEPTS you will learn in this chapter include:

- Why intravascular devices are an important cause of systemic bloodstream infections
- How to minimize the risk of nosocomial infections related to intravascular devices
- How to insert, care for and remove intravenous lines
- How to set up and administer blood or blood products

BACKGROUND

The use of intravascular devices, both venous and arterial, to deliver sterile fluids, medications and nutritional products, as well as for central monitoring of blood pressure and other hemodynamic functions, has dramatically increased during the past decade. It is estimated that about 50% of all patients admitted to hospitals will receive intravenous therapy, creating a large population at risk for local and systemic blood stream infections.

Because catheters inserted into the venous or arterial bloodstream bypass the normal skin defense mechanism, these devices provide a way for microorganisms to enter the bloodstream from:

- the device at the time of insertion,
- subsequent contamination of the device or attachments (e.g., tubing connected to the blood monitoring apparatus or the fluids being administered), or
- pathogens on the skin surrounding the insertion site.

The risk of infection associated with the use of intravascular devices can be reduced by following recommended infection prevention practices related to their insertion (e.g., the use of aseptic technique) and by better management of the device once it is in place. In many countries, poor infection prevention practices, such as infrequent handwashing or use of antiseptic handrub, and the improper use of gloves often result in increased rates of local and systemic infections. Moreover, when intravascular devices (e.g., central venous catheters) are introduced in hospitals where laboratory services to provide identification and antimicrobial susceptibility testing are lacking or inadequate, the treatment

of life-threatening bloodstream infections stemming from these devices is often unsuccessful or results in the emergence of resistant organisms.

This chapter provides guidelines for the preparation, insertion and maintenance of common intravascular devices (i.e., peripheral venous lines for the administration of fluids, electrolytes and blood or blood products).[1]

DEFINITIONS

- **Exit site infection (microbiologic diagnosis)**. Clinical infection in which culture of the discharge (pus or fluid) at the exit site yields a microorganism, with or without microbiologic evidence of bloodstream infection.
- **Phlebitis**. Area of swelling, redness, warmth and tenderness of the skin around the site where the intravascular catheter comes out of the skin (the exit site). If phlebitis is associated with other signs of infection, such as fever and pus coming from the exit site, it is classified as a **clinical exit site infection**.
- **Pocket infection**. Infected fluid isolated from the area around a totally implanted intravascular device, with or without microbiologic evidence of bloodstream infection.
- **Tunnel infection**. Tenderness, redness and swelling for more than 2 cm (about 1 inch) along the tract of an intravascular catheter, with or without microbiologic evidence of local or bloodstream infection.

EPIDEMIOLOGY AND MICROBIOLOGY

Peripheral venous catheters, if inserted using recommended infection prevention practices, are rarely (less than 1%) associated with systemic (bloodstream) infections (**Table 24-1**). If they are not properly maintained, however, these devices can cause local reactions (e.g., phlebitis) that potentially increase the risk of subsequent infection. By contrast, nontunneled central venous pressure catheters (CVCs) account for nearly 90% of all catheter-related bloodstream infections, with the remaining due to use of the other devices (Maki 1992). Because nosocomial bloodstream infections have a relatively high morbidity compared to other types of nosocomial infections, in the range of 10–20% (CDC and HICPAC 1996), it is extremely important that where possible, midline catheters, which have lower rates of phlebitis and infection, be used rather than nontunneled CVCs (Garner and HICPAC 1996).

[1] Insertion and maintenance of other devices (e.g., peripheral artery catheters and nontunneled or tunneled central venous lines) require personnel with special training to minimize the risk of catheter-related complications (e.g., pneumothorax) or infections (CDC and HICPAC 1996). If IV teams are not available, insertion and removal should be the responsibility of a few well-trained staff members using aseptic techniques. Even for insertion of peripheral venous catheters, students or unskilled or inexperienced staff should be directly supervised, and the number of attempts should be limited for patient safety and comfort.

Table 24-1. Types of Intravascular Devices and Comments on Their Use

Peripheral venous catheter	Usually inserted into the veins of the forearm or the hand; most commonly used short-term intravascular device; rarely associated with bloodstream infection
Peripheral arterial catheter	For short-term use; commonly used to monitor hemodynamic status and to determine blood gas levels of critically ill patients; risk of bloodstream infection may approach that of CVCs[1]
Midline catheter	Peripheral catheter (size 7.6–20.3 cm or 3–8 inches) is inserted via the antecubital fossa (forearm) into the proximal basilic or cephalic veins, but it does not enter central veins; is associated with lower rates of phlebitis and infection than CVCs
Nontunneled CVC	Most commonly used CVC; accounts for an estimated 90% of all catheter-related bloodstream infections; increased risk of infection with internal jugular vein site of insertion
Pulmonary artery catheter	Inserted through a Teflon introducer and typically remains in place for an average duration of only 3 days; most catheters are heparin-bonded to reduce catheter thrombosis and microbial adherence to the catheter
Pressure-monitoring system	Used in conjunction with arterial catheter; associated with both epidemic and endemic nosocomial bloodstream infections; source is often the fluid column in the tubing between the patient's intravascular catheter and the pressure-monitoring apparatus, contaminated infusate, or contaminated nondisposable transducers
Peripherally inserted central catheter	Provides an alternative to subclavian or jugular vein catheterization; is inserted via a peripheral vein into the superior vena cava, usually by way of cephalic and basilar veins; is easier to maintain and is associated with fewer mechanical complications (e.g., hemothorax) than are nontunneled CVCs
Tunneled CVC	Surgically implanted CVC with tunneled portion exiting the skin and a Dacron cuff just inside the exit site; the cuff inhibits migration of organisms into the catheter tract by stimulating growth of the surrounding tissue, thus sealing the catheter tract; used to provide long-term vascular access to patients
Totally implantable device	A subcutaneous port or reservoir with self-sealing septum is tunneled beneath the skin and is accessed by a needle through intact skin; low rates of infection

[1] CVC: central venous catheter

Adapted from: Mermel et al 2001.

Most infections are caused by contamination of the catheter with organisms from the patient's skin or the health worker's hands during insertion, with the catheter providing a direct path to the bloodstream. Once the catheter is inserted, pathogens can be transferred into the bloodstream in four ways:

1. by traveling along the catheter-tissue interface,
2. through contamination of the hub,
3. through contaminated infusion fluid, and
4. through the bloodstream from another site of infection.

Microbiology

Both gram-negative bacteria and staphylococci are primary causes of catheter-related infection; however, with the advent of the HIV/AIDS epidemic, infections with fungi are increasingly being reported (Jarvis and Hughes 1993). Some microorganisms, especially coagulase-negative *Staphylococcus aureus* and pseudomonas and acinetobacter species, adhere to the fibrin film that forms on the inside wall of catheters within days after insertion. As a consequence**,** infection with these organisms is quite common, especially if the infection occurs within 10 days of insertion (Raad et al 1993). For devices left in place longer than 30 days (e.g., tunneled CVCs), bloodstream infections are more likely due to the contamination of the hub of the catheter, especially if frequent handling of the hub occurs (Schaberg, Culver and Gaynes 1991).

RISK FACTORS

A number of factors increase the risk of infection from intravascular devices. For example, infection rates are higher among patients in large hospitals who may be especially ill, those with burns or surgical wounds or those who are malnourished or immunocompromised (e.g., by HIV/AIDS or chronic corticosteroid treatment). In addition, the rates are higher for certain devices (e.g., nontunneled CVCs), the type of fluid being infused (parenteral nutritional products are most risky) and the length of time the catheter is left in place (Jarvis et al 1991; Maki and Mermel 1998; Mayhall 1992).

Contaminated equipment and solutions also provide microorganisms with a way to get into the bloodstream. The following device-related factors increase the risk of infection:

- **Before insertion**:
  - Cracks in infusion bottles
  - Punctures in plastic containers
  - Contaminated infusion fluid or additives
  - Leaky IV administration sets with multiple connections
  - Unsterile preparation of intravenous infusion fluid

- **During use**:
  - Multiple changes of IV fluid containers while using the same IV administration set

- Multiple injections and irrigations of the system
- Central venous pressure measurement apparatus

Person-to-person contact also increases the risk of infection associated with intravascular devices. These include:

- Cross-contamination with other infected areas of the patient's body either by the patient or on the hands of the health worker.
- Cross-contamination from another patient via the hands of the health worker.
- Cross-contamination from the patient when the health worker comes in contact with the patient's blood during insertion, care of the insertion site or removal of the catheter.
- Poor insertion or dressing change technique.

REDUCING THE RISK OF NOSOCOMIAL INFECTIONS

All Types of Intravascular Devices

Hand Hygiene and Gloves

- Wash hands before touching any of the IV set components. (If hands are visibly clean, you can disinfect them with an antiseptic handrub made from 60–90% ethyl or isopropyl alcohol and an emollient, such as glycerin.)
- Clean examination gloves or reprocessed high-level disinfected surgical gloves should be put on just before touching the insertion site or the hub of the needle or catheter.
- Wash hands or use a waterless, alcohol-based antiseptic handrub after removing gloves.

Site Care and Dressings

- If the site for inserting the catheter is visibly dirty, wash it with soap and clean water and dry it before applying the skin antiseptic.

Note: PVI releases free iodine (the active antiseptic agent) slowly.

- If using povidone-iodine (PVI) as the antiseptic agent, allow it to dry after applying or wait at least 2 minutes before insertion.
- Applying antimicrobial ointment around the insertion site does not reduce the risk of infection (APIC 2002).
- Transparent, adherent dressings allow inspection of the site, act as tape to hold the catheter or needle, and may be more comfortable, but they are expensive and there is no evidence, based on randomized controlled trials, that they reduce the risk of infection compared to sterile or clean gauze and surgical tape.
- Dressings can be left in place for up to 72 hours if they are kept dry. (They should be changed immediately if they get wet, soiled or loose.)

- Gauze and tape dressings need to be changed if an inspection of the site is necessary.
- The catheter or needle site should be gently palpated daily for tenderness.
- The insertion site should be inspected if the patient develops tenderness or fever without an obvious cause (CDC and HICPAC 1996).

Peripheral Catheters (Venous and Arterial)

Site Selection and Rotation

- For adults, hand veins are preferred over arm veins, and arm veins over leg and foot veins. (Needles and catheters inserted in leg and foot veins are more likely to cause inflammation at the insertion site or phlebitis.)
- Rotating sites at 72–96 hours will reduce phlebitis and local infection. (Teflon or polyurethane catheters are preferred over steel needles because they are less apt to perforate the vein with movement.)
- If only short-term (less than 48 hours) IV infusion is planned, straight or butterfly needles are less irritating than plastic catheters and have lower rates of infection.
- Because straight and butterfly needles frequently infiltrate, they should not be used with solutions that could cause tissue necrosis.
- Inline filters, except for administering blood or blood products, are not recommended; they are more expensive, less effective and more prone to cause infusion problems than if the solutions are filtered in the pharmacy after preparation (CDC and HICPAC 1996).

Central Venous Catheters

Site Care and Dressings

Note: In 1994, Raad et al reported lower rates of blood stream infections when full barrier precautions were used compared to insertions by staff using only sterile gloves and a small drape with a hole in the center.

- If the site for inserting the catheter is visibly dirty, wash it with soap and clean water and dry it before applying the skin antiseptic.
- Use 2% chlorhexidine gluconate, 10% PVI or 60–90% alcohol for skin prep. (In 1991 Maki, Ringer and Alvarado reported that the infection rate with chlorhexidine was 84% lower than with PVI or alcohol.)
- Insertion should be done using full barrier precautions (sterile or high-level disinfected gloves, gown, mask and site drape) in a procedure area, not at the bedside.

Changing Fluids and Infusion (Administration) Sets

- Change infusion bottles or plastic bags with parenteral solutions every 24 hours.
- Change infusion bottles or plastic bags with lipid emulsion given alone within 12 hours (CDC and HICPAC 1996).

- Infusion (administration) sets (including piggybacks) should be changed whenever they are damaged and at 72 hours routinely. (If the tubing becomes disconnected, wipe the hub of the needle or plastic catheter with 60–90% alcohol and connect it to a new infusion set.)
- Tubing used to administer blood, blood products or lipid emulsions should be replaced within 24 hours (CDC and HICPAC 1996).

INSERTION, MAINTENANCE AND REMOVAL OF PERIPHERAL VENOUS LINES

Insertion Procedure for Establishing an Intravenous (IV) Line

STEP 1: Make sure all items are available:

- IV solution bag or bottle
- Straight or butterfly needle or plastic catheter (steel needle inserter covered with soft plastic tubing that is left in place after the needle is withdrawn)
- Infusion (administration) set—infants and children require drip rate (drips per mL) and volume control devices
- Antiseptic solution (e.g., 2% chlorhexidine, 60–90% alcohol or 10% povidone-iodine) and sterile or clean gauze squares (2 x 2 or cotton swabs)
- Surgical tape or transparent dressing
- Clean tourniquet
- Clean or new arm board
- Towel to place under patient's hand or forearm
- IV pole
- Clean pair of examination gloves (If examination gloves are not available, reprocessed high-level disinfected surgical gloves can be used.)
- Basin of clean warm water, soap, face cloth and clean dry towel
- Plastic bag or leakproof, covered waste container for disposal of contaminated items

Note: Use distal veins (farthest from the wrist or elbow) first and avoid placing the IV line over the wrist or in the patient's dominant hand (the one s/he writes with).

STEP 2: Explain the procedure to the patient.

STEP 3: Prior to starting the procedure, identify the best vein(s) for inserting IV needle or plastic catheter.

STEP 4: If the venipuncture site is visibly soiled, first wash it with soap and clean water and dry with a clean cloth.[1]

[1] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

STEP 5: Wash hands with soap and clean water and dry with a clean, dry towel or air dry. (Alternatively, if hands are not visibly soiled, apply 5 mL, about 1 teaspoonful, of an antiseptic handrub to both hands and vigorously rub hands and between fingers until dry.)

STEP 6: Check the IV solution (bottle or plastic bag) to be sure it is correct and the right additives, such as potassium, have been added.

STEP 7: Open the infusion set and assemble parts, if necessary using aseptic technique (e.g., don't touch ends of tubing).

STEP 8: Insert infusion set into solution bottle or bag:

- Remove protective cover from solution bottle or bag without touching the opening.
- Remove protective cap covering insertion spike without touching the spike and insert spike into stopper of IV bottle or opening of IV bag.

STEP 9: Fill infusion tubing:

- Compress drip chamber and release.
- Remove protective cover of IV tubing and release roller clamp to allow fluid to fill the tubing, close the roller clamp and replace the protective cover. (Check to be sure tubing is clear of air bubbles.)

STEP 10: With forearm and hand hanging down, place tourniquet 10–12 cm (5–6 inches) above the insertion site. (Ask patient to open and close fist and/or tap lightly over the vein to make it easier to see or feel.)

STEP 11: With tourniquet in place and vein filled, place hand and arm on the clean towel on bed or on arm board.

STEP 12: Put clean examination gloves on both hands.

STEP 13: Cleanse insertion site with antiseptic solution using a circular motion outward from the insertion site. (If using povidone-iodine, allow it to dry, about 2 minutes, because it only releases free iodine, the active antiseptic agent, slowly).

STEP 14: Attach straight or butterfly needle or plastic catheter to a syringe if blood is to be taken for testing. If not, the needle or butterfly should be attached to sterile end of the IV tubing.

Note: Do not insert unattached needle or catheter into a vein and allow blood to drip out on the patient's hand, forearm, the bed or floor!

STEP 15: Fix the vein by placing the thumb over the vein and gently pulling against the direction of insertion.

STEP 16: Insert needle or catheter with the bevel up using the dominant hand. Look for blood return in the tubing and carefully advance the needle or butterfly until the hub rests at the venipuncture site. (With catheters, after getting blood return, advance the needle about 1 cm (½ inch), withdraw the inner insertion needle and then advance the plastic catheter to the hub.)

STEP 17: While stabilizing the needle or catheter, release the tourniquet and roller clamp to permit a rate of flow sufficient to keep the IV line open.

Note: The tourniquet should be washed with soap and water, rinsed and dried whenever visibly soiled and wiped with 0.5% chlorine solution or 60–90% alcohol between patients.

STEP 18: Secure the needle or catheter by placing a narrow piece of tape (1 cm or ½ inch) under the hub with the adhesive side up and cross tape it over the hub. Then place a second piece of narrow tape directly across the hub of the needle or catheter.

STEP 19: Place a sterile gauze square (2 x 2) over the venipuncture site and secure with two pieces of tape. (Alternatively, place a transparent dressing over the venipuncture site.)

STEP 20: Prior to removing gloves, place any blood-contaminated waste items (cotton or gauze squares) in a plastic bag or leakproof, covered waste container.

STEP 21: Remove gloves by inverting and place them either in a plastic bag or waste container.

STEP 22: Wash hands or use antiseptic handrub as above.

Note: Carefully write the date and time of placement of the IV line and needle size on the dressing.

STEP 23: Secure the wrist or forearm to the arm board by applying two strips of tape directly across wrist or forearm. (To minimize discomfort when removing the arm board, attach a shorter piece of tape to the longer piece—adhesive side to adhesive side—that will cover the wrist or arm.)

STEP 24: Adjust the flow rate to the correct number of drips per minute.

Maintenance of IV Line

STEP 1: Observe patient hourly to determine her/his response to the fluid therapy and check that:

- IV line is open and running (if a straight or butterfly needle is being used, check for infiltration);
- correct amount of fluid is being infused; and
- proper flow rate (drops per minute) is maintained.

Note: If only short-term (less than 48 hours) IV infusion is planned, straight or butterfly needles are less irritating than plastic catheters and have lower rates of infection.

STEP 2: Check every 8 hours for phlebitis or evidence of infection.

STEP 3: Rotate the infusion site at 72–96 hours, when practical, to reduce the risk of phlebitis and local infection.

STEP 4: The infusion (administration) sets (including the piggybacks) should be changed whenever they are damaged and at 72 hours routinely.

STEP 5: If the tubing becomes disconnected, wipe the hub of the needle or the plastic catheter with 60–90% alcohol and connect to a **new** infusion set.

Changing IV Solutions

STEP 1: Prepare to change the solution when about 50 mL remains in the bottle or bag.

STEP 2: Check to be sure the drip chamber is half full.

STEP 3: Wash hands or use antiseptic handrub as above.

STEP 4: Prepare the new solution. If using a plastic bag, remove the protective cover from the entry site. If using a glass bottle, remove the metal cap, metal disk and rubber disk. Do not touch the entry site on the bag or bottle.

STEP 5: Move the roller clamp to stop the flow.

STEP 6: Remove the old solution from the IV pole.

STEP 7: Remove the spike from the old IV solution bag or bottle, and without touching the tip, insert the spike into the new IV solution bag or bottle.

STEP 8: Hang the new bag or bottle and discard the empty bag or bottle according to hospital policy.

STEP 9: Check for air in the tubing.

STEP 10: Make sure the drip chamber is half full.

STEP 11: Regulate the flow to the prescribed rate.

STEP 12: Observe the patient hourly to determine her/his response to the fluid therapy and check that:

- the IV line is open and running (if a straight or butterfly needle is used, check for infiltration);
- the correct amount of fluid is being infused; and
- the proper flow rate (drops per minute) is maintained.

STEP 13: Check every 8 hours for phlebitis or evidence of infection.

Changing IV Tubing

STEP 1: Determine that a new infusion set is needed if:

- there is a puncture of the infusion tubing;
- the tubing becomes contaminated;
- the tubing becomes blocked (e.g., after an infusion of packed red blood cells, whole blood or albumin); or
- the date on the dressing indicates the tubing has been in place 24 hours if used to administer blood, blood products or lipid emulsions, or 96 hours for other fluids.

STEP 2: Make sure the following items are available:

- Plastic bag or a leakproof, covered waste container for disposing of contaminated items
- Infusion tubing

STEP 3: If a new IV dressing must be applied, additional items are:

- Sterile or clean gauze squares (2 x 2) and surgical tape or sterile, wide (1 inch) bandaid
- Antiseptic solution (2% chlorhexidene gluconate, 60–90% alcohol or 10% povidone-iodine)
- Alcohol swabs
- Clean pair of examination gloves (If examination gloves are not available, reprocessed high-level disinfected surgical gloves can be used.)

STEP 4: Wash hands or use antiseptic handrub as above.

STEP 5: Open a new infusion set and assemble it if necessary.

STEP 6: Partially open a sterile gauze square package and place it on the bed near the IV puncture site.

STEP 7: Move the roller clamp to the "off" position on the old infusion tubing, remove the insertion spike from the IV fluid bag or bottle and hang the end of the old IV tubing over the IV pole.

STEP 8: Quickly remove the protective cap on the insertion spike of the new infusion tubing and insert it into the entry site of the IV infusion bottle or bag.

STEP 9: Compress and release the drip chamber to fill half full.

STEP 10: Open the roller clamp, remove the protective cap from the needle adapter, allow the tubing to completely fill, move the roller clamp to the "off" position and replace the protective cap without touching the tip.

STEP 11: Put clean examination gloves on both hands.

STEP 12: If the needle or catheter hub is not visible, carefully remove the IV dressing and place it in a plastic bag or leakproof, covered waste container.

STEP 13: Stabilize the hub of the IV needle or plastic catheter, gently twist and pull out the old tubing, quickly remove the protective cap from the needle adapter of the new tubing, and insert the tubing into the hub of the needle or plastic catheter.

STEP 14: Open the roller clamp on the new tubing and adjust the rate of flow as ordered.

STEP 15: Discard the old tubing in a plastic bag or leakproof, covered waste container.

STEP 16: If necessary, apply a new dressing by placing a gauze square (2 x 2) over the venipuncture site and secure it with two pieces of tape. (Alternatively, place a transparent dressing over the venipuncture site.)

STEP 17: Remove gloves by inverting and place them either in a plastic bag or waste container.

STEP 18: Wash hands or use antiseptic handrub as above.

IV Removal Procedure

STEP 1: Make sure all items are available:

- Clean pair of examination gloves (If examination gloves are not available, reprocessed high-level disinfected surgical gloves can be used.)
- Antiseptic solution (2% chlorhexidene gluconate, 60–90% alcohol or 10% povidone-iodine)
- Gauze squares (2 x 2) and surgical tape or a sterile, wide (1 inch) bandaid
- Puncture-resistant sharps container within arm's reach if a straight or butterfly needle was used
- Plastic bag or leakproof, covered waste container for disposing of the contaminated items

STEP 2: Wash hands or use antiseptic handrub as above.

STEP 3: Stop the infusion by closing the roller clamp.

STEP 4: Put clean examination gloves on both hands.

STEP 5: Remove the arm board and dressing and discard it in a plastic bag or leakproof, covered waste container.

STEP 6: Check the patient's hand or wrist for phlebitis or evidence of an infection.

STEP 7: Carefully remove the needle or the plastic catheter with one hand and with the other hand cover the insertion site with a sterile gauze square (2 x 2).

STEP 8: Press firmly for about a minute, or alternatively place two pieces of narrow tape, about 1 cm or ½ inch wide, directly across the gauze square.

STEP 9: Alternatively, after pressing on the gauze square, remove it and cover the insertion site with a sterile bandaid.

STEP 10: Prior to removing gloves, discard the needle or plastic catheter in a sharps container and place the IV tubing and any blood-contaminated waste items (cotton or gauze squares) in a plastic bag or leakproof, covered waste container.

STEP 11: Remove gloves by inverting and place them either in a plastic bag or a leakproof, covered waste container.

STEP 12: Wash hands or use antiseptic handrub as above.

ADMINISTERING BLOOD OR BLOOD PRODUCTS

Transfusion Procedure

STEP 1: Make sure all items for starting an IV (**Step 1** above), are available.

STEP 2: Additional items needed include:

- A #18 or #19 straight or butterfly needle or a plastic catheter
- An infusion (administration) set that has an inline filter, and the tubing also should be Y-type
- A 250 or 500 mL sterile, isotonic (0.9%) saline solution bottle or bag

Note: Sterile saline solution prevents red blood cells from breaking (hemolysis) and is used to keep the IV line open before starting blood or blood products, between units of blood or blood products, and after completing the transfusion to flush the inline filter and infusion tubing.

STEP 3: Explain the procedure to the patient; determine if s/he has ever had a transfusion and note reactions, if any.

STEP 4: Ask the patient to report chills, headaches, itching or rash immediately.

STEP 5: Establish an IV line with a large-gauge (#18 or #19 straight or butterfly needle or plastic catheter) as detailed in **Insertion Procedure for Establishing an IV line** (**Steps 3** through **21** above).

Note: Patients who have had blood transfusion reactions may have greater fear of transfusion, and may be at increased risk of recurrence.

STEP 6: Keep the IV line open with a sterile 0.9% (isotonic) saline solution.

STEP 7: With another health worker, correctly identify the blood product and make sure you have the correct patient:

- Confirm the patient's name and check her/his armband if available.
- Check the compatibility tag attached to the blood bag, including the expiration date of the blood (after which it should not be used).
- For whole blood, check the ABO group and Rh type, which should be on the patient's chart.
- Double-check the blood or type of blood product with the physician's order.
- Check the blood for clots.
- Record the baseline pulse and blood pressure.

STEP 8: Remove the protective cover from the blood or blood products bag or the bottle without touching the opening.

STEP 9: If using a Y-type infusion set, remove the protective cap covering the second insertion spike without touching the spike and insert it into the blood bag or bottle. (If using a single tubing infusion set, carefully remove the insertion spike from the saline bag or bottle, and without touching the spike, insert it into the blood bag or bottle.)

STEP 10: Begin the transfusion:

Note: If a reaction is suspected, **stop** the transfusion, flush the line with isotonic saline and infuse slowly to keep the IV line open and notify the blood bank or transfusion service and physician.

- Fill the inline filter.
- Adjust the rate to 2 mL per minute.

STEP 11: Immerse both gloved hands in a 0.5% chlorine solution, remove gloves by inverting and place them in the plastic bag or a leakproof, covered waste container.

STEP 12: Wash hands or use antiseptic handrub as above.

STEP 13: Monitor the patient's vital signs:

- Take the pulse and blood pressure every 5 minutes for the first 15 minutes of the transfusion and hourly thereafter.
- Observe the patient for flushing (red face or cheeks), itching, difficulty breathing, hives (clear fluid-filled lesions on the skin) or any other rash when checking the vital signs.

STEP 14: Record the administration of the blood or blood product in the patient's chart.

STEP 15: When the transfusion is completed, exchange a new IV solution bottle or bag for the empty blood bag or bottle and return it to the blood bank.

STEP 16: If no further infusions are ordered:

- Remove the needle or plastic catheter and infusion set as detailed in **IV Removal Procedure** (**Steps 1** through **9** above).
- Return the blood bag or bottle and tubing to the blood bank.

STEP 17: Prior to removing gloves, discard the needle or plastic catheter in a sharps container and place the blood administration kit, IV tubing and any blood-contaminated waste items (cotton or gauze squares) in a plastic bag or leakproof, covered waste container.

STEP 18: Remove gloves by inverting and place them either in a plastic bag or waste container.

STEP 19: Wash hands or use antiseptic handrub as above.

REFERENCES

Association for Professionals in Infection Control and Epidemiology (APIC). 2002. Intravascular device infections, in *APIC Text of Infection Control and Epidemiology*, revised ed. APIC: Washington, DC, pp 30-1–30-8.

Centers for Disease Control and Prevention (CDC) and Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guidelines for prevention of intravascular device-related infections. *Infect Control Hosp Epidemiol* 17(7): 438–473. (Authors: Pearson ML and HICPAC).

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80 and *Am J Infect Control* 24(1): 24–52.

Jarvis WR and JM Hughes. 1993. Nosocomial gastrointestinal infections, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Williams & Wilkins: Baltimore, MD, pp 708–745.

Jarvis WR et al. 1991. Nosocomial infection rates in adult and pediatric intensive care units in the United States. *Am J Med* 91(Suppl 3B): 185S–191S.

Maki DG and LA Mermel. 1998. Infections due to infusion therapy, in *Hospital Infections*. JV Bennett and PS Brachman (eds.). Lippincott-Raven: Philadelphia, pp 698–724.

Maki DG. 1992. Infections due to infusion therapy, in *Hospital Infections,* 3rd ed. JV Bennett and PS Brachman (eds). Little, Brown and Company: Boston, pp 849–898.

Maki DG, M Ringer and CJ Alvarado. 1991. Prospective, randomized trial of povidone-iodine, alcohol and chlorhexidine for prevention of infection associated with central venous and arterial catheters. *Lancet* 338(8763): 339–343.

Mayhall CG. 1992. Diagnosis and management of infections of implantable devices used for prolonged venous access. *Curr Clin Top Infect Dis* 12: 83–110.

Mermel LA et al. 2001. Guidelines for the management of intravascular catheter-related infections. *Infect Control Hosp Epidemiol* 22(4): 222–242.

Raad I et al. 1994. Prevention of central venous catheter-related infections by using maximal sterile barrier precautions during insertion. *Infect Control Hosp Epidemiol* 15(4 pt 1): 231–238.

Raad I et al. 1993. Ultrastructural analysis of indwelling vascular catheters: A quantitative relationship between luminal colonization and duration of placement. *J Infect Dis* 168(2): 400–407.

Schaberg DR, DH Culver and RP Gaynes. 1991. Major trends in the microbial etiology of nosocomial infection. *Am J Med* 91(Suppl 3B): 72s–75s.

TWENTY-FIVE

PREVENTING MATERNAL AND NEWBORN INFECTIONS

KEY CONCEPTS you will learn in this chapter include:

- What the special features of maternal and newborn infections are
- Why maternal and newborn infections are more common in developing countries
- How prevention can decrease the risk of many fetal and newborn infectious diseases
- How to decrease the risk of maternal and newborn infections following labor and delivery

BACKGROUND

In no other area of primary healthcare is the disparity between developed and developing country morbidity and mortality greater than for pregnant women and their newborns. For example, in some of the poorest countries, maternal mortality rates are a hundred times higher than those in Western Europe and the United States.

In developed countries, most pregnant women are healthy and well nourished. They deliver their babies in a hospital or birthing center, and only a few are subjected to the wide variety of invasive and diagnostic procedures experienced by most other hospitalized patients. Even for those having cesarean sections, the surgery is short (i.e., usually less than an hour) and usually uncomplicated. Urinary catheterization, if required, is brief (1–2 days) and rarely is assisted ventilation required postoperatively. Thus, the risk of nosocomial (hospital-acquired) infection, or infection with a multidrug-resistant organism following delivery, even after cesarean section, is low compared with other types of hospitalized patients. In fact, were it not for the nearly five-fold increase in cesarean section rates from 5.5% in 1978 to 30% during the early 1990s, maternal morbidity and mortality would be even lower.[1] Finally, because most women in developed countries start attending prenatal clinics early (i.e., first trimester) and are fully immunized, the risk of serious infection to the fetus and newborn is low as well.

The situation in countries with limited resources, however, is radically different in nearly every aspect. In these countries, anywhere from 50–80%

[1] Mortality from cesarean section still remains at least two to four times higher than that following vaginal delivery (Petitti et al 1982).

of pregnant women give birth at home—usually alone or with a family member—and most have received only limited antenatal care. They are poorly nourished and anemic. If a complication occurs in labor requiring cesarean section, they usually arrive at the hospital too late, when they are near death. Moreover, even if they survive the surgery the rate of postoperative infection is high (15–60%), and wound infections, the most serious complication, are very common. Added to this in recent years is the fact that in some countries up to 30% of pregnant women are seropositive for HIV. This, coupled with the resurgence of tuberculosis, especially drug-resistant strains, further complicates the situation. As a consequence, pregnant women in developing countries are at much higher risk for acquiring a nosocomial infection following delivery than their counterparts in developed countries.

Newborns do not fare well either! Other than maternal tetanus toxoid immunization during pregnancy, and treatment to prevent congenital syphilis, few other preventive measures to protect the fetus and newborn are routinely available. For example, with the exception of prenatal HIV testing and antiretroviral treatment in a few countries, screening and treatment for other infectious diseases (e.g., gonorrhea and chlamydia) are not available because of the cost and lack of laboratory capability. Moreover, in Africa and parts of Asia, malaria is a major problem that can adversely affect pregnancy outcome. Thus, in countries where healthcare resources are limited, little progress has been made over the past decade in preventing fetal and newborn diseases, and improving the quality and availability of newborn services in hospitals has been slow as well.

DEFINITIONS

- **Endometritis**. Acute postpartum infection of the lining (endometrium) of the uterus with extension into the smooth muscle wall (myometrium). Clinical features include fever, usually developing on the first or second postpartum day, uterine tenderness, lower abdominal pain, foul-smelling vaginal discharge (lochia) and signs of peritonitis in women who have had a cesarean section.
- **Episiotomy**. Surgical cut made in the perineum (usually at the 6 o'clock position) just prior to delivery. The purpose is to facilitate delivery of the presenting part and minimize the risk of injury to the perineal area. Episiotomies, however, are associated with increased bleeding, may lead to increased tearing (3rd or 4th degree perineal laceration), can become infected and, most importantly, usually are not necessary.
- **Intra-amniotic infection syndrome (IAIS), also referred to as amnionitis or chorioamnionitis**. Acute detectable infection in the uterus and its contents (fetus, placenta and amniotic fluid) during pregnancy. It occurs in a small percentage (<5%) of term pregnancies, but in up to 25% of women with preterm labor (before 37 weeks gestation). It is usually related to colonization of the uterine cavity

with organisms present in the cervix and vagina after prolonged ruptured membranes and labor. In cases of IAIS associated with serious, and often fatal, newborn infection and postpartum endometritis, the most common organisms isolated from amniotic fluid are group B streptococci and *E. coli*.

- **Invasive group B streptococcal sepsis**. Newborn infection characterized by bacteremia, pneumonia, meningitis and death in up to 25% of infants with the infection. It occurs most commonly following IAIS. Other sites of infection include newborn skin infections (cellulitis) and infections in bones (osteomyelitis).
- **Nosocomial infection in newborns**. Infection occurring after birth but excluding those infections known to have been transmitted across the placenta such as congenital syphilis, cytomegalovirus, rubella, varicella (chicken pox) and the protozoan parasite, *Toxoplasmosis gondii*.
- **Nosocomial infection in obstetrical patients**. Infection that is neither present nor incubating at the time the patient is admitted to the hospital. Most urinary tract infections and endometritis are nosocomial even though the causative organism may be endogenous (i.e., present in the maternal lower genital tract prior to delivery).
- **Septic pelvic thrombophlebitis**. Thrombosis (blockage) of the deep pelvic veins due to inflammation and blood clots. It is uncommon (approximately 1 in 2000 deliveries). Predisposing factors include cesarean section after long labor (>24 hours), premature rupture of membranes, difficult delivery (forceps or vacuum extraction), anemia and malnutrition.

EPIDEMIOLOGY

Maternal Infections

In developing countries, postpartum infection remains second only to postpartum hemorrhage as a cause of maternal deaths and is the leading cause of serious maternal complications of childbirth. This is still the case despite the fact that more than 150 years have elapsed since Semmelweis and Holmes independently determined not only that childbed fever, puerperal sepsis, was spread from woman to woman on the hands of physicians, but also that outbreaks of this deadly disease could be prevented by:

- rigorously enforcing handwashing with chlorinated lime before delivery, and
- boiling all instruments and utensils after use when treating an infected postpartum woman.

Employing these preventive efforts, Holmes reported a dramatic decrease in maternal mortality from 16% to 1% (Holmes 1843).

In many countries, acute endometritis (puerperal sepsis) is still the most common postpartum infection. Rates of infection range from a low of 1–3% following vaginal deliveries in hospitals with high quality services and excellent infection prevention practices, to as high as 85% following high-risk cesarean sections in poorly nourished, exhausted patients who have their operation in large teaching hospitals that have limited healthcare resources (Hemsell 1991).

Cesarean section is the most important factor contributing to both the frequency and severity of postpartum endometritis (Gibbs 1980). For example, patients who have cesarean sections are at least 10 times more likely to become infected than patients who deliver vaginally (Minkoff and Schwarz 1980). Moreover, patients undergoing their first (primary) cesarean section are at an even greater risk for an infection or other complications compared to patients having an elective, repeat section.

The distribution and type of nosocomial infections following cesarean section in the US are shown in **Table 25-1**. While organ/space surgical site infections such as endometritis account for over half, the most serious and costly are wound infections (nearly 20%). For example, patients with wound infections typically spend 7 days longer in the hospital than those without infection and 4 days longer than patients with endometritis. Wound infections are primarily the result of direct contamination of the incisional area with organisms in the endometrial cavity at the time of surgery. Predisposing factors for wound infection are women who:

- have bacterial vaginosis (*Gardnerella vaginalis)* isolated from the endometrium,
- have a cesarean section during the second stage of labor, or
- had infection of the fetal membranes (chorioamnionitis) diagnosed prior to delivery (Mead 1993).

Table 25-1. Distribution of Nosocomial Infections in Cesarean Section

Incisional Surgical Site Infection	Organ/Space Surgical Site Infection[1]	Urinary Tract Infection	Pneumonia	Primary Bloodstream Infection	Other
19%	55%	12%	3%	2%	9%

[1] Primarily endometritis; may also include infections such as intra-abdominal abscess.

Adapted from: Horan et al 1993.

Other obstetrical infections are less frequent, ranging from less than 1% to 15%. In decreasing order of frequency these include:

- Nosocomial urinary tract infections (about 12% and largely in women who had a cesarean section)

- Episiotomy infections (<5%, usually simple and uncomplicated)
- Nosocomial pneumonia (3% and almost always in post cesarean section patients)
- Septicemia (2% and largely in post cesarean section patients)
- Breast infection (mastitis) in postpartum nursing women (<3%)

Maternal nosocomial infection rates in most developing countries, however, are considerably higher.

Fetal and Newborn Infections

Fetal and newborn infections are classified based on whether they were acquired *in utero* (transplacentally), during passage through the birth canal (vertical transmission) or in the neonatal period (i.e., during the first 28 days following birth).

In utero infections include those caused by:

- viruses—cytomegalovirus, rubella, varicella (chicken pox/zoster), HIV and parovirus;
- protozoa—*toxoplasmosis gondii*; and
- bacteria—congenital syphilis.

Intrapartum (mother to newborn) and immediate postpartum newborn infections include those caused by:

- viruses—hepatitis B, hepatitis C, HIV, herpes simplex virus (HSV), human papillomavirus (HPV) and parovirus; and
- bacteria—*E. coli,* group B streptococci, yeast (candida species); conjunctivitis due to chlamydia, gonorrhea or *Listeria monocytogenes,* and a number of infections due to gram-negative anaerobic bacilli.

In addition, a number of other organisms that can colonize and sometimes infect newborns during the first month of life include:

- viruses—cytomegalovirus, enterovirus, respiratory syncytial virus and rhinovirus;
- protozoa—malaria in many tropical countries; and
- bacteria—tuberculosis and tetanus.

Strictly speaking, only newborn infections acquired during passage through the birth canal or in the neonatal period are considered nosocomial. Determining whether an infection is nosocomial or was present or incubating prior to admission to the hospital is extremely difficult—and often not useful. For example, a common definition of nosocomial intra-amniotic infection syndrome (IAIS) is one that occurs

following either an invasive action (e.g., vaginal examination or intrauterine fetal monitoring) or an attempt to induce labor more than 24 hours previously. Using this definition, less than 1% of IAISs would be considered nosocomial in most hospitals (Mead 1993)!

MICROBIOLOGY

Causes of Maternal Infections

Most postpartum infections are caused by endogenous flora—microorganisms that are normally present in the genital tract but usually cause no disease until labor, delivery or postpartum. Nearly 30 bacteria have been identified as being present in the lower genital tract (vulva, vagina and cervix) at any time (Faro 1990). While some of these, including several fungi, are considered nonpathogenic under most circumstances at least 20, including *E. coli, S. aureus, Proteus mirabilis* and *Klebsiella pneumoniae*, are pathogenic.

The organisms most commonly isolated from women with endometritis are listed in **Table 25-2**. Because endometrial and urine cultures may be misleading due to the contaminating vaginal and cervical flora, not surprisingly postpartum women with clinical evidence of endometritis or urinary tract infections are cultured less frequently than patients with other types of infections (Mead 1993).

Table 25-2. Commonly Isolated Organisms in Women with Endometritis

AEROBES

- Gram-positive cocci
  - Group B streptococcus
  - Group A streptococcus
  - Enterococcus
  - Streptococcus sp. (other)
  - Staphylococcus sp.
- Gram-negative
  - *Escherichia coli*
  - *Klebsiella pneumoniae*
  - *Proteus mirabilis*

ANAEROBES

- Gram-positive cocci
  - Peptococcus sp.
  - Peptostreptococcus sp.
- Gram-positive bacilli
  - Clostridium sp.
- Gram-negative bacilli
  - *Prevotella bivia*
  - *Bacteroides fragilis*
  - Bacteroides sp. (other)

Adapted from: Cox and Gilstrap 1989.

Colonization and Infection in Newborns

Most infants are delivered from a sterile environment inside the uterus. During and after birth, however, they are rapidly exposed to numerous microorganisms that colonize their skin, nasopharynx and gastrointestinal tract. Sick newborns, subjected to multiple invasive procedures (e.g., endotracheal tubes or umbilical artery catheters), may be colonized at multiple sites with numerous other organisms, particularly gram-negative bacteria.

The skin of the newborn is a major initial site of bacterial colonization, particularly for *Staphylococcus aureus,* which is most often acquired from within the nursery rather than from the mother. Any break or cut in the skin provides an opportunity for infection to develop with this pathogenic organism. In addition, at birth the newborn has at least one open surgical wound (the umbilicus) that is highly susceptible to infection. A circumcision, if performed, is another, and if a fetal scalp electrode was used during labor, then the newborn has a third site as well. Therefore, to minimize the risk of infection in the newborn period, all sites must be cared for using aseptic technique.

Although severe infection in a full term infant is uncommon, when it occurs it often is secondary to group B streptococci, *E. coli, L. monocytogenes, Citrobacter diversus,* salmonella, chlamydia, herpes simplex virus (HSV) or enteroviruses. All of these organisms can be transmitted to other infants in the nursery on the hands of hospital staff unless Standard Precautions are strictly followed, especially those for handwashing (or use of antiseptic handrub) and gloves.

PREVENTING FETAL AND NEWBORN INFECTIOUS DISEASES

Prevention has long been the only viable alternative in the fight against most of the devastating fetal and newborn infectious diseases such as congenital rubella, cytomegalovirus, varicella (chicken pox), syphilis, toxoplasmosis and tetanus. And, over the past 50 years, preventive efforts have successfully reduced the risk of serious fetal and newborn infections in developed countries. This success has been accomplished through:

- maternal immunization (tetanus, rubella, varicella and hepatitis B);
- antenatal treatment of maternal syphilis, gonorrhea and chlamydia;
- prophylactic use of postnatal eye drops to prevent chlamydial, gonorrheal and yeast (candida) eye infections (conjunctivitis);
- prophylactic treatment of pregnant women at risk of group B streptococcal disease; and most recently
- maternal (antenatal and intrapartum) and newborn (postnatal) treatment with antiretroviral (ARV) drugs to prevent HIV.

In countries with limited healthcare resources, however, little progress has been made in preventing these fetal and newborn infectious diseases with the exception of neonatal tetanus and syphilis.

In **Appendix K,** specific information regarding prevention of the most important fetal and newborn infectious diseases is presented. In addition, infection prevention guidelines are provided that are designed to minimize the risk of transmission to other newborns, postpartum mothers and susceptible health workers and other staff.

REDUCING THE RISK OF MATERNAL AND NEWBORN INFECTIONS

In this section, guidelines are provided for reducing the risk of maternal and newborn infections during and following either vaginal or cesarean delivery. Basic information also is included on managing outbreaks in newborn nurseries and neonatal intensive care units (NICUs). Simple, preventive practices that can be used in all settings and by all healthcare workers are described.

Because of the increasing risk of exposure to HIV and other bloodborne viruses during labor, delivery and resuscitation of the baby, if required, health workers also should be protected. The conscientious use of Standard Precautions, especially handwashing and use of gloves, face shields and plastic or rubber aprons, can minimize these risks; therefore, the appropriate use of personal protective equipment is emphasized throughout this chapter.

Minimizing the Risk of Infection during Labor and Vaginal Delivery

Babies are born in a variety of settings around the world, especially when the birth is a normal delivery. Although vaginal delivery does not require the aseptic conditions of an operating room, a few simple practices can make the procedure safer for the mother, the infant and the healthcare provider. For example, using the "three cleans" approach—keeping the hands, perineal area and umbilical area clean during and following childbirth—and having clean delivery kits help improve the safety of home births for both mother and newborn.

Vaginal deliveries are associated with a number of factors that increase a woman's risk of endometritis or urinary tract infection. These include:

- prolonged ruptured membranes (>24 hours),
- trauma to the birth canal (vaginal or perineal lacerations and urethral tears),
- manual removal of the placenta due to retained placenta or placental fragments,
- episiotomy, and
- midforceps delivery (Hemsell 1991; Newton, Prihoda and Gibbs 1990).

Each of these provides a means for microorganisms to enter, or to be placed inside, the uterus (uterine cavity). While the first three factors can happen regardless of where the birth occurs (at home or in the hospital), the last two are related solely to deliveries occurring in hospital maternity units. Moreover, when babies are born in a hospital or healthcare facility, another factor that increases the risk of maternal infection is vaginal examinations, especially those performed by medical and midwifery students. For example, in one study it was found that the risk of endometritis was 27% if seven or fewer vaginal examinations were performed but rose to 71% when more than seven were performed (Iffy et al 1984).

To minimize this risk:

- Use a clean pair of examination gloves, or high-level disinfected surgical gloves that have been reprocessed, for each examination. (Sterile gloves are not necessary for vaginal examinations.)
- Avoid pushing the tip of the examining finger up against the opening to the cervix (cervical os) until active labor occurs or until the decision has been made to induce labor.
- Carefully limit cases for student training to those patients in active and progressive labor.

Vaginal Delivery (Maternity Unit of Birthing Center)

Steps that can be taken to decrease the risk of maternal infection **before** and **during** delivery include:

STEP 1: Make sure the following items are available:

- Two pairs of high-level disinfected or sterile surgical gloves
- Pair of high-level disinfected or sterile "fingerless" surgical gloves (**Chapter 7**)
- Pair of clean examination gloves for washing the perineum
- Basin of clean warm water, soap, a face cloth and clean dry towel[2]
- Plastic or rubber apron and face shield (or a mask and goggles)
- Waterless, alcohol-based antiseptic handrub or antiseptic solution (e.g., 2% chlorhexidine gluconate or 10% povidone-iodine)
- High-level disinfected or sterile blunt scissors (Mayo)
- High-level disinfected or sterile cord clamp or cloth to tie off the cord
- Injectable oxytocin (with or without methergine) or oral misoprostol

[2] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to removes particulate matter (if necessary), or use chlorinated water—water treated with dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

- High-level disinfected or sterile urinary catheter (straight, rubber or metal) and clean basin to collect urine (optional)
- Package of gauze squares
- Clean basin for the placenta
- Clean drape or cloth for wrapping the baby
- Clean perineal pads
- Light source (a flashlight or lamp) if needed
- Puncture-resistant sharps container (within arm's reach if possible)
- Plastic bucket with a tight fitting lid, filled with 0.5% chlorine solution for decontamination
- Plastic bag or a leakproof, covered waste container for disposal of contaminated waste items

If episiotomy is required, the following will be needed as well:

- High-level disinfected or sterile needle holder
- High-level disinfected or sterile tissue forceps
- #O chromic suture on, or with, a curved, minimally blunt (preferred) or cutting suture needle
- Local anesthetic (without epinephrine)

Prior to Delivery

STEP 2: Once the patient is positioned for delivery, put examination gloves on both hands and wash the perineal area (vulva, perineum, and anal region) with soap and clean water[3]:

- Use a downward and backward motion when washing the perineal area so that fecal organisms will not be introduced into the vagina.
- Clean the anal area last and place the washcloth or towel in a plastic container.

Shaving perineal (pubic) hair increases the risk of infection associated with delivery (Landry and Kilpatrick 1997).

STEP 3: Immerse both gloved hands in a 0.5% chlorine solution, remove gloves by inverting, and place them in the plastic bag or leakproof, covered waste container.

[3] Use of antiseptic solutions for cleaning the perineal area has not been shown to decrease postpartum infections in mother or baby (AAP and ACOG 1997).

STEP 4: Thoroughly wash hands, especially between the fingers, and forearms up to the elbows with soap and clean water and dry with a clean, dry towel or air dry.

STEP 5: Apply 5 mL (about 1 teaspoonful) of the antiseptic handrub to hand and forearms and rub until dry; repeat application and rubbing 2 more times for a total of at least 2 minutes, using a total of about 15 mL (3 teaspoonfuls) of the handrub. (If handrub is not available, apply an antiseptic solution to hands and forearms, rinse with clean water and dry hands.)

Note: If reprocessed high-level disinfected or sterile surgical gloves are used, double gloving is recommended to reduce the risk of exposure to blood or other body fluids.

STEP 6: Put high-level disinfected or sterile surgical gloves on both hands.

STEP 7: Wear protective equipment including a plastic or rubber apron and face shield (or a mask and goggles) because splashing of blood and blood-tinged amniotic fluid can be expected.

Note: Shoe covers, unless they are resistant to fluids, are not helpful.

During Delivery

- If resuscitation of the infant is required, use mechanical suction if available. (If mouth suction of the airway cannot be avoided, place a trap in the line.)
- If manual removal of the placenta is required, fingerless surgical gloves should be used to avoid contaminating the forearm with blood. To use fingerless gloves:

    - First, remove the surgical glove from one or both hands using the technique described in **Chapter 4**.
    - Next, put on a fingerless high-level disinfected or sterile surgical glove(s) and pull up onto the forearm(s) using the technique described in **Chapter 7**.
    - Finally, put a new high-level disinfected or sterile surgical glove on one or both hands.

After Delivery

STEP 8: Before removing gloves, put the placenta in the clean basin and place all waste items (e.g., blood-stained gauze) in the plastic bag or leakproof, covered waste container.

STEP 9: If an episiotomy was done or there were vaginal or perineal tears requiring surgical repair:

- Place sharps (suture needles) in the puncture-resistant sharps container.
- If disposing of hypodermic needle and syringe, hold the needle under the surface of a 0.5% chlorine solution, fill the syringe and push out (flush) three times; then put in a puncture-resistant sharps container.

Alternatively, if reusing syringe (and needle), fill syringe with needle attached with 0.5% chlorine solution and soak for 10 minutes for decontamination.

STEP 10: Immerse both gloved hands in a 0.5% chlorine solution; remove gloves by inverting, and place in the plastic bag or leakproof, covered waste container if discarding them. If reusing them, place them in a 0.5 % chlorine solution for 10 minutes for decontamination.

STEP 11: Wash hands or use an antiseptic handrub.

Minimizing the Risk of Infection During Cesarean Section

Cesarean sections should be performed using the same standards as for any general surgical procedure as described in **Chapter 7**. Certain features that make this operation different are:

- The surgeon and assistant should wear a face shield (or mask and goggles) and a plastic or rubber apron over their scrub suits because splashing of blood and blood-tinged amniotic fluid can be expected.
- Double gloving is recommended, especially if reprocessed sterile or high-level surgical gloves are used.
- A first or second-generation cephalosporin should be given intravenously after the cord is clamped if the section is high risk (i.e., prolonged ruptured membranes or labor of any duration). (See **Table 23-2** for details.)[4]
- The health worker receiving the infant should wash her/his hands and put on clean examination gloves (or reprocessed high-level disinfected surgical gloves) before handling the baby.
- The baby should be placed on a clean towel after being passed off to the health worker caring for the infant.
- Change surgical gloves before manually removing the placenta. (If available, use elbow-length surgical gloves or a combination of fingerless gloves and a new pair of surgical gloves as described in **Chapter 7** and above.)
- With prolonged ruptured membranes or with documented intra-amniotic infection syndrome (chorioamnionitis):
  - Avoid spillage of amniotic fluid into the abdominal cavity.
  - Place folded, moistened sterile laparotomy pads or towels on either side of the uterus (paracolic gutters) to catch as much contaminated amniotic fluid as possible.
  - If large amounts of meconium or amniotic fluid spill into the abdominal cavity, remove the laparotomy pads or towels in the

[4] Several well-designed studies have demonstrated that intravenous (IV) antibiotic prophylaxis reduces the risk of endometritis by about 50% after nonelective cesarean sections. (Cunningham et al 1983; Padilla, Spence and Beauchamp 1983). Antibiotic lavage of the abdominal cavity, however, offers no advantage over IV administration, is time-consuming and has been shown in one study to be less effective (Conover and Moore 1984).

gutters and lavage the cavity with sterile isotonic (0.9%) saline solution.
- Do not explore the peritoneal cavity unless absolutely necessary, and then only after closure of the uterine incision and surgical gloves have been changed.

- If the cervix is closed and membranes were not ruptured prior to the cesarean section:

  - Dilate the cervix from below (i.e., through the vagina) sufficiently to permit the outflow of blood and fluid (lochia) after delivering the baby and placenta.
  - Insert the gloved finger into the cervix only once to dilate it.
  - Do not go back and forth or remove the hand from the pelvis and then put the finger back into the cervix.
  - When dilation is completed, remove the gloves and put on a new pair of sterile or high-level disinfected surgical gloves (**Chapter 4**).

- To minimize postoperative wound infections:

  - Patients should not be shaved prior to surgery. (If it is necessary to remove pubic or abdominal hair, clip the hair with scissors just prior to surgery.)
  - Make the skin incision with a scalpel rather than with electrocautery.
  - After the fascia is closed, irrigate the wound with sterile isotonic (0.9%) saline and then blot it dry.
  - Whenever possible, do not place drains in the subcutaneous layer.
  - Close the skin edges using a subcuticular technique.
  - Apply a sterile dressing and care for the wound as described in **Chapter 23.**

Aseptic technique is broken whenever a nonsterile area is touched, such as when the gloved hand reaches down into the pelvis to extract the baby's head or buttocks. Whenever a sterile or high-level disinfected surgical glove (or gloves) becomes contaminated, it should be changed as soon as possible (see **Chapter 4** for how to change gloves).

Postpartum Care of the Mother

Minimizing the risk of nosocomial infection in mothers during the postpartum period includes the following:

- Wear examination or utility gloves when handling perineal pads, touching lochia (vaginal discharge) or touching the episiotomy.

- In the immediate postpartum period, check to be sure she is voiding without difficulty.
- Teach her how to wash the perineal area with boiled water after changing a pad or having a bowel movement (defecation).
- If the patient is breastfeeding, teach her how to care for her breasts and nipples to avoid infection (mastitis).
- If delivery was by cesarean section, to avoid pulmonary problems during the immediate postoperative period and for the next few days:
    - use pain medication cautiously,
    - encourage her to move about in bed and take deep breaths frequently, and
    - get her out of bed and walking within the first 12 hours (**Chapter 27**).
- If delivery was by cesarean section and an indwelling catheter was inserted, to avoid urinary problems:
    - check to be sure urine is flowing and the urine collection system is intact,
    - follow the "Tips for Preventing Infections" in **Chapter 22**, and
    - remove the catheter as soon as possible (within 24–48 hours).

Postnatal Care of the Newborn

Minimizing the risk of nosocomial infection in the newborn involves the following:

- Wear gloves and plastic or rubber apron when handling the infant until blood, meconium or amniotic fluid has been removed from the infant's skin.
- Careful removal of blood and other body fluids using a cotton cloth, not gauze, soaked in warm water followed by drying the skin may minimize the risk of infection.
- Wash hands before holding or caring for the infant. Alternatively, a waterless, alcohol-based antiseptic handrub can be used.
- Bathing or washing the newborn should be delayed until the baby's temperature has stabilized (usually about 6 hours). The buttocks and perineal areas are the most important to keep clean. They should be washed after each diaper change using a cotton cloth soaked in warm soapy water, and then carefully dried.
- Cover gowns or masks are not required when handling infants.
- No single method of cord care has proved to be better in preventing infection. General suggestions are:
    - Wash hands, or use an antiseptic hand rub, before and after cord care.
    - Keep the cord stump clean and dry.

- Do not cover the cord stump with a dressing or bandage.
- Fold the diaper below the cord stump.
- If the cord stump gets soiled or dirty, gently wash it with boiled soapy water, and rinse with boiled water and dry with a clean cloth.
- Explain to the mother that if the cord stump becomes red or is draining pus or blood she should bring the baby to a clinic or hospital equipped to care for newborns as soon as possible.

Management of Outbreaks in the Nursery or NICU

A presumptive epidemic in a nursery or neonatal intensive care unit (NICU) is defined as finding two or more newborns with the same condition (e.g., skin infection or infectious diarrhea) at the same time. If an epidemic or outbreak of a particular disease such as diarrhea is suspected, the first step is to assess it promptly and carefully to:

- determine the need for laboratory or epidemiologic studies (if available);
- identify the source of the diarrhea (e.g., patients, staff or visitors) and the means of transmission (e.g., contamination via hands of staff, parents or visitors); and
- decide on the type of control measures required to prevent the spread of the infection. (See **Chapter 28** for details of how to conduct an outbreak investigation.)

Even if an intensive investigation is not required, the control measures (e.g., strict isolation or placing all infected newborns in a common area) should be monitored to be sure that they have been effective and the problem is resolving.

For additional information on management of outbreaks due to:

- infectious diarrhea and foodborne infections, see **Chapter 26**; and
- specific airborne, droplet or contact diseases, see **Chapter 21** and **Appendix I**.

In addition, management of infected newborns, based on their presumptive diagnosis (clinical findings), is described in **Appendix K**.

REFERENCES

American Academy of Pediatrics (AAP) and American Academy of Obstetricians and Gynecologists (ACOG). 1997. *Guidelines for Perinatal Care*, 4th ed, revised. AAP: Elk Grove, IL.

Conover W and TR Moore. 1984. Comparison of irrigation and intravenous antibiotic prophylaxis at cesarean section. *Obstet Gynecol* 63(6): 787–791.

Cox S and L Gilstrap. 1989. Postpartum endometritis. *Obstet Gynecol Clin North Am* 16(2): 363–371.

Cunningham FG et al. 1983. Perioperative antimicrobials for cesarean delivery: Before or after cord clamping? *Obstet Gynecol* 62(2): 151–154.

Faro S. 1990. *Infections in Pregnancy.* Gilstrap L and S Faro (eds). John Wiley & Sons, Inc.: New York.

Gibbs RS. 1980. Clinical risk factors for puerperal infection. *Obstet Gynecol* 55(Suppl 5): 178S–183S.

Hemsell DL. 1991. Prophylactic antibiotics in gynecologic and obstetric surgery. *Rev Infect Dis* 13(Suppl 10): S821–S841.

Holmes OW. 1843. The contagiousness of puerperal fever. *N Engl Q J Med Surg* 1: 503–530.

Horan T et al. 1993. NNIS data: Nosocomial infections in surgical patients in the United States—January 1986–June 1992. *Infect Control Hosp Epidemiol* 14(2): 73–80.

Iffy L et al. 1984. Infection control in obstetrics, in *Operative Perinatology: Invasive Obstetric Techniques.* Iffy L and C Charles (eds). Macmillan: New York, pp 86–99.

Landry K and D Kilpatrick. 1997. Why shave a mother before she gives birth? *Matern Child Nurs* 2: 189–190.

Mead PB. 1993. Prevention and control of nosocomial infections in obstetrics and gynecology, in *Prevention and Control of Nosocomial Infections.* 3rd ed, revised. Wenzel RP (ed). Williams & Wilkins. Baltimore, MD, pp 776–795.

Minkoff HL and RH Schwarz. 1980. The rising cesarean section rate: Can it be safely reversed? *Obstet Gynecol* 56(2): 135–143.

Newton ER, TJ Prihoda and RS Gibbs. 1990. A clinical and microbiologic analysis of risk factors for puerperal endometritis. *Obstet Gynecol* 75(3 Pt 1): 402–406.

Padilla SL, MR Spence and PJ Beauchamp. 1983. Single-dose ampicillin for cesarean section prophylaxis. *Obstet Gynecol* 61(4): 463–466.

Petitti DB et al. 1982. In-hospital maternal mortality in the United States: Time trends and relation to the method of delivery. *Obstet Gynecol* 59(1): 6–12.

TWENTY-SIX

PREVENTING INFECTIOUS DIARRHEA AND MANAGING FOOD AND WATER SERVICES

KEY CONCEPTS you will learn in this chapter include:

- What the impact of nosocomial diarrhea is on healthcare
- What the common causes of infectious diarrhea are
- How to manage food and water services in healthcare facilities
- How to prevent nosocomial outbreaks of foodborne diarrhea
- How to prepare clean and safe drinking water

BACKGROUND

Nosocomial diarrhea is a common problem in hospitals, children's care facilities and nursing homes (Lynch et al 1997). While not listed as one of the commonest nosocomial infections, recent studies suggest that nosocomial diarrhea may occur more frequently than reported and, in fact, may be the most common site of nosocomial infection in some types of healthcare facilities. The cost and morbidity also are greater than one would expect because diarrhea often is not reported or studied as a nosocomial infection (Farr 1991).

Controlling the spread of nosocomial diarrhea from contaminated food is an ongoing concern in hospitals and nursing homes. Frequently this is due to poorly trained food handling staff using unsafe practices involving the storage, preparation and handling of raw meat, chicken, fish and fresh eggs as well as some vegetables. Moreover, because the quality of drinking water in countries with limited resources often is unsafe, preventing patients getting diarrhea from drinking contaminated water and controlling outbreaks of waterborne infectious diarrhea are persistent problems. Therefore, in this chapter guidelines are provided for reducing the risk of nosocomial diarrhea from contaminated food or water by improving sanitation, food handling and staff hygiene. In addition, practical suggestions for managing outbreaks of infectious diarrhea in the newborn nursery or neonatal intensive care unit (NICU) are summarized in this chapter, and more detailed guidance and instructions are supplied in **Chapter 28**. Finally, information is included describing how to prepare and maintain a continuous supply of clean and safe water for drinking and medical use.

DEFINITIONS

- **Clean water**. Natural or chemically treated and filtered water that is safe to drink and use for other purposes (e.g., handwashing and medical instrument cleaning) because it meets specified public health standards. These standards include: zero levels of microorganisms, such as bacteria (fecal coliform and *E. coli*), parasites (*Giardia lamblia*) and viruses (hepatitis A or E); low turbidity (cloudiness due to particulate matter and other contaminants); and minimum levels of disinfectants, disinfectant by-products, inorganic and organic chemicals and radioactive materials. At a minimum, clean water should be free of microorganisms and have low turbidity (is clear, not cloudy).
- **Endemic illness or disease**. Infectious disease, such as cholera, which is continuously present at some level (prevalence) in a particular country or region.
- **Environmental hygiene**. Process of maintaining a clean, healthy and pleasing patient and work environment.
- **Epidemic**. Rapid spread of an infectious disease, such as cholera, among many individuals in a hospital or community at the same time.
- **Nosocomial diarrhea**. On at least 2 consecutive days having at least three loose or watery stools with the onset more than 72 hours after admission to the hospital (or more days than the incubation period if the agent is known).

EPIDEMIOLOGY

In hospitalized patients, the incubation period for infectious diarrhea due to various bacterial and viral organisms may be shorter due to decreased immunity or other risk factors. Although infectious diarrhea is definitively diagnosed only when the bacterial or viral agent is identified, many cases of diarrhea are never fully diagnosed. In the US, nosocomial diarrhea has been reported to vary from less than 1 per 100 admissions among children to over 30 per 100 admissions for elderly adults (McFarland 1993). In developing countries, because diarrhea is so common, even reasonable estimates are lacking.

Infectious agents causing diarrhea are transmitted by the fecal/oral route in a number of ways:

- by eating or drinking contaminated food or fluids;
- from a patient handling contaminated articles (e.g., with feces), and then putting her/his hands in the mouth;
- from the contaminated hands of health workers; and
- from contaminated medical instruments (e.g., gastroscopes) that enter the gastrointestinal (GI) tract.

Noninfectious diarrhea is usually caused by medications such as antibiotics or procedures such as endoscopy, nasogastric feeding or X-ray studies using barium and enemas. While noninfectious diarrhea is a common problem, usually it does not last long and does not require treatment.

The most serious of the infectious GI illnesses is neonatal necrotizing enterocolitis that kills the cells of the gut and leads to peritonitis and septicemia. This condition primarily affects premature infants and outbreaks have occurred in NICUs and special care nurseries. Although no specific agent has been identified, the epidemiology suggests that a transmissible agent (unknown bacteria or virus) is the cause.

MICROBIOLOGY

Outbreaks of diarrhea in hospitals, nursing homes and NICUs have been associated with a wide variety of organisms including salmonella, shigella, *Clostridium difficile*, vibrio (cholera), *Candida albicans*, *Staphylococcus aureus*, cryptosporidium, rotavirus and other enteroviruses. Some of the most common bacterial and viral agents causing infectious diarrhea, their incubation period and most prominent clinical characteristics are listed below:

Common Bacterial Agents

- ***Salmonella*** (salmonellosis) produces fever, nausea and vomiting followed by diarrhea that frequently contains mucus (whitish and stringy), but rarely blood, in stools. The incubation period is less than 72 hours (3 days) when large doses of organisms are eaten in contaminated food or drinks. Outbreaks among children, however, are commonly the results of contact transmission, and about 50% of exposed infants will develop illness once a case is introduced in a nursery. Adults, on the other hand, usually get salmonellosis from contaminated food, drinks or inadequately cleaned and disinfected medical instruments such as endoscopes. Once several patients or staff are infected, transmission by contact to new susceptibles may be very rapid. Salmonellosis is a common cause of infectious diarrhea, accounting for more than 50% of all diarrhea outbreaks in nursing homes in which the causative agent was identified (Levine et al 1991).

  Control of outbreaks may be difficult; some nurseries or wards have had to restrict new admissions. Safe food handling is essential for prevention, especially raw (uncooked) eggs or egg products (e.g., homemade mayonnaise or tartar sauce). Antibiotic treatment prolongs the time the infected person may carry the organism in her/his GI system, but antibiotic treatment may be necessary for septic or severally ill patients. For most, use of oral rehydration solution (ORS) and supportive care are sufficient.

- ***Shigella*** (shigellosis) produces rapid onset of diarrhea, with stools containing mucus and often blood. Infected persons are often more sick than is typical for other infecting agents. The incubation period is 1–6 days, and the usual source is fecal/oral transmission from acutely

infected patients. Outbreaks are less common than with salmonella or viral agents, and patients shed the organisms only for a short period after becoming symptomatic. Antibiotic treatment may be needed, but use of ORS is most important.

- ***Clostridium difficile*** (formerly called antibiotic-resistant diarrhea or pseudomembranous colitis) has increasingly become an important cause of diarrhea. It may be the cause of nearly half of all cases of nosocomial diarrhea in adult hospitalized patients (McFarland 1995). The diarrhea ranges from mild and self-limiting to severe pseudomembranous colitis, which can be fatal. Because *C. difficile* is present in the stools of infants and preschool children, colonization without clinical disease apparently occurs. Its presence in the GI tract gradually decreases with age. In addition, *C. difficile* may become endemic in the nursery and other high-risk units. No nosocomial outbreaks have been associated with foodborne transmission, suggesting that contact (person to person) transmission from contaminated articles or the hands of staff is responsible. For example, one report noted that when culture-negative patients were placed in a hospital room currently or previously occupied by a person with *C. difficile* diarrhea, they were more likely to develop this type of diarrhea than patients placed in rooms where no patient had had *C. difficile* diarrhea (McFarland et al 1989). This suggests the organism can persist on inanimate articles (e.g., lamps, door handles or bed rails) for some time unless rooms are thoroughly cleaned between patients.
- ***Escherichia coli*** strains that cause acute diarrhea have not been reported to be nosocomially transmitted. Toxic strains have been transmitted in restaurants from contaminated meat that was not cooked sufficiently to kill the organisms and could be a problem in healthcare facilities that prepare their own meals from raw meat.
- ***Vibrio cholerae*** subgroups produce acute, severe diarrheal disease characterized by local outbreaks, widespread epidemics and occasional individual outbreaks. Cholera is usually associated with contaminated water sources (see last section of this chapter for detailed information on prevention). Treatment usually consists of ORS.

Common Viral Agents

- ***Rotavirus*** cause sudden onset of vomiting and diarrhea within 48-72 hours (2–3 days) after exposure. Fever and upper respiratory symptoms are present in about half the cases. In addition the virus may be present in the sputum or secretions for several days. This may account for the extremely rapid transmission and seasonal peak in infections during winter. Symptoms subside in a few days, but the stool may contain virus for up to 2 weeks. Rotaviruses are the most common cause of diarrhea in children under five. Because it is highly infectious, during nursery outbreaks nearly all infants will become infected. Like *C. difficile*, the virus survives well on inanimate surfaces and may become endemic in hospitals.

- ***Norwalk and similar viruses*** cause sudden onset of diarrhea, nausea, vomiting, mild fever and abdominal cramps for about 24 hours. The incubation period is short, a few days. These viruses are usually associated with food (salad, raw vegetables and shellfish) and waterborne contamination, but nosocomial outbreaks can occur suggesting that person-to-person transmission does occur.

RISK FACTORS

Host risk factors for nosocomial diarrhea include young age; old age; patients with burns, trauma or decreased immunity; decreased gastric acidity; and altered flora in the stomach and gut, such as that occurring with antibiotic treatment in some people. Health worker risk factors include lack of hand hygiene, especially in food handlers, and noncompliance with glove use.

REDUCING THE RISK OF NOSCOMIAL DIARRHEA

Hand Cleanliness and Gloves

Enteric organisms (e.g., *E. coli* and rotavirus) are transferred to susceptible people via the hands of health workers and patients who get the organisms on their hands from direct contact with feces or indirectly from articles that have fecal material on them. To reduce the risk of exposure and cross-contamination:

Remember: Wash hands, or use an antiseptic handrub, before eating, drinking or smoking.

- Patients and staff should wash their hands or apply waterless, alcohol-based antiseptic handrub after contact with fecal organisms in bathrooms, on toilet articles such as bedpans, or on patients who have fecal incontinence.
- Wear new, clean examination gloves before touching mucous membranes (mouth or nose) of all patients, including infants and children.
- Utility or heavy-duty gloves should be worn if activities are likely to involve touching or handling feces (e.g., changing the bed of a patient who has fecal incontinence).

Environmental Contamination and Soiled Linen

- Clean and wipe bedpans and bathroom equipment that are regularly handled by patients and staff with a disinfectant (0.5% chlorine solution or 1% Lysol) daily and whenever they have been used.
- When fecal soilage does occur (e.g., incontinent patients or diaper accidents) all soiled articles should be immediately cleaned and disinfected.
- Staff who sort linen should wear utility or heavy-duty gloves. Also, soiled linen should be bundled so that leakage does not occur, and all linen should be handled as if fecal contamination is present (see **Chapter 13** for details).

- Wear gloves when handling linen soiled with moist body substances, used diapers or toilet paper, and place in a plastic bag or leakproof, covered waste container.

Food Service Personnel

- Routine stool cultures of food service staff are ineffective, expensive and provide no benefit. Even chronic carriers may only shed organisms intermittently. For example, routine nasal (anterior nares) cultures only identify 10–30% of chronic carriers of *Staphylococcus aureus* linked to food poisoning.
- Food handlers with diarrhea should be immediately removed from handling foods. They should not return to food handling or work with immunocompromised patients or intensive care or transplant patients until all symptoms are over for 24–48 hours.

Patients with Diarrhea

Patients with diarrhea from any cause should be managed according to Standard Precautions with Transmission-Based Precautions added if the diagnosis indicates (see **Chapter 21**). Other precautions include moving roommates to another room in the hospital if fecal contamination is likely, encouraging staff to use gowns or plastic aprons for clean-up activities, and providing frequent cleaning of articles that might be contaminated. If available, plastic-backed diapers for infants, children and even adults should be used. Infants born to mothers with diarrhea should not enter the regular nursery. Rather, rooming-in should be provided for mother and infant, and the mother should be taught good hygiene.

Outbreak Management

The successful management of outbreaks of diarrhea related to common source contamination in healthcare facilities usually requires a number of procedures:

- finding the common source and eliminating it,
- grouping patients with diarrhea together and not allowing the sharing of equipment or staff with any new or uninfected patients,
- discharging affected and unaffected patients early if their care can be managed at home,
- making sure that housekeeping is thorough and frequently performed, and
- providing separate space and extra staff to care for affected infants in the case of nursery or neonatal ICU outbreaks (see **Chapter 28** for details).

Because management of outbreaks is expensive, preventing them by eliminating the risks of food- or waterborne infections is more cost-effective.

MANAGING FOOD AND WATER SERVICES

Nosocomial transmission of fecal organisms by contaminated food or water can be reduced considerably by improving sanitation, food handling

and staff hygiene, including handwashing or the use of waterless, alcohol-based handrub products.

Factors that increase risk of nosocomial diarrhea in hospitals include the fact that they:

- serve food for more hours than restaurants,
- serve ill and immunocompromised patients,
- must transport and distribute food at greater distances, and
- prepare nasogastric (enteral) feedings and special diets.

In addition, staff often are transient, poorly trained and may have other health problems that can contribute to poor quality food services.

The most common organisms associated with foodborne outbreaks include salmonella and *Clostridium difficile*. *Shigella*, *Escherichia coli*, hepatitis A virus (HAV) and cholera species are next in frequency. For foodborne infection to occur:

- food and fluids must be infected (contain sufficient numbers of infectious organisms),
- preparation and storage processes and practices must allow organisms to persist, and
- food must be served to susceptible hosts.

In a study of 162 foodborne outbreaks in hospitals and nursing homes in the US, the errors associated with the outbreaks were as follows:

- 38% stemmed from improper holding temperatures for prepared food,
- 18% were due to poor hygiene of a presumably infected employee,
- 14% were caused by improper cooking,
- 13% were due to contaminated equipment,
- 5% occurred because food was obtained from an unsafe source, and
- 9% were caused by other factors (Villarino et al 1992).

Food Service Guidelines

All activities in the food service department should be monitored at regular intervals to be sure that safety standards are being followed, including:

Holding temperatures should be above 60°C/140°F or below 7°C/45°F. Thermometers for food storage should be checked periodically. Warm, perishable foods should be cooled before being stored, or stored in shallow containers so that the center temperatures are not warm enough for bacteria to thrive on or for toxins to be produced—both are common causes of staphylococcal "food poisoning" outbreaks.

Cooking should be complete. All parts of the item should reach the proper temperature. In particular, frozen meats should be thawed before cooking to avoid the presence of cold spots in the interior. If there is any suspicion that the interior temperature is below the proper level at the end of cooking, it should be checked by taking a thermometer reading.

Personal health and hygiene of food service staff are of great importance and should be supervised by a knowledgeable person. Food handlers should have convenient access to handwashing facilities and be provided with individual containers of waterless antiseptic handrub if possible (**Chapter 3**). A handwashing station should have access to clean water, soap and a clean towel (single-use or disposable towels are preferable). If common towels are used, they should be changed when visibly soiled and at least every 4 hours. Because food service employees may not appreciate the importance of handwashing, it must be reinforced in staff training and through appropriate behavior modeled by supervisors and managers. Staff need to know:

- basic principles of personal hygiene and how good hygiene helps prevent disease transmission;
- importance of reporting GI problems or skin lesions, especially on the hands;
- how to properly inspect, prepare and store the foods they handle;
- how to clean and operate equipment they use such as slicers, blenders and dishwashers; and
- waste management.

Ensure equipment cleaning and disinfection, especially cutting boards used for preparing raw meat, fish or poultry. These can become contaminated and need to be cleaned and disinfected between uses. Other equipment should be cleaned daily. A system for inspecting and maintaining all equipment in central kitchens is important.

Purchase raw food from known vendors that meet local inspection standards, if possible. Foods prepared at home should not be shared with other hospitalized patients. Also, perishable food brought from home must be consumed immediately, and any leftovers returned home with the visitors.

Note: If refrigeration is not available, prepared formula should be used within 4 hours. Even if refrigeration is available, formula should not be held more 24 hours (ADA 1991).

Contaminated powdered infant formula is also a problem. In one study of 141 powdered breast milk substitutes from 28 countries, more than half (52%) of the samples contained gram-negative organisms (Muytjens, Roelofs-Willemse and Jaspar 1988). Most problems with infant formulas arise during preparation and storage of the freshly prepared product. Formula should be prepared in a clean space where no other work is being done at the time. The container should be clean and dry, and water used

for preparing the formula should be boiled vigorously for 5 minutes. The prepared formula should be placed in clean bottles, which have been rinsed with boiled water and allowed to dry.

Preparation of Clean Water

Water boiled for 1–5 minutes is considered safe to drink, while water boiled for 20 minutes is high-level disinfected. Alternatively, water can be disinfected and made safe for drinking by adding a small amount of sodium hypochlorite (commercial bleach) solution.[1] For example, 15 mL (0.5 ounces) of a 1% solution will disinfect 20 liters (21 quarts) of water, leaving a residual chlorine level sufficient to protect the water for 24 hours (CDC 2000). Chlorination should be done just before storing the water in a container, preferably one with a narrow neck. (Storage containers often become contaminated if the neck is large enough to permit hands or utensils to enter.)

The preparation of clean water containing up to 0.001% (10 ppm) sodium hypochlorite solution is inexpensive, easy to do and often is needed during emergency situations (e.g., during floods or other natural disasters that may lead to contamination of the water system). In addition, being able to prepare clean water daily is important in healthcare facilities, such as small health clinics located in rural or remote areas. Frequently, they do not have access to a reliable water source that can be used for handwashing and for cleaning instruments, surgical gloves and other medical items prior to final processing.

A Sustainable Source of Clean Water

Recently, portable systems have become commercially available that generate up to 0.6 % (6000 ppm) sodium hypochlorite from common table salt, contaminated water from rivers, shallow wells or ponds, and electricity. The power source may be either 110/220V A/C, D/C or from solar photovoltaic cells. These systems are designed to operate in remote or rural areas under extremely harsh conditions for many years. For example, a small system, operating on solar energy, can treat up to 20,000 liters (over 21,000 quarts) of contaminated water per day in only 8 hours (ESE 2002). These systems are relatively inexpensive (about $1,500 for a small system), and extremely easy to use and maintain. They require nothing more than occasionally putting the electrode in vinegar (3–5% acetic acid) to dissolve phosphates and carbonates that gradually build up on the hypochlorite-generating electrode's cathode (negative pole). Moreover, they provide a sustainable source of clean and safe drinking water or a continuous supply of sodium hypochlorite for medical use (e.g., decontamination or chemical HLD of instruments).

[1] If tap water is cloudy, most particulates (debris and organic material) can be removed by filtering through four layers of moderately woven cotton cloth, such as cheese cloth or old sari material, before boiling or treating with dilute chlorine (sodium hypochlorite) solution (Colwell et al 2003; Huq et al 1996).

How to Prevent the Spread of Cholera

In a number of countries, such as Bangladesh, cholera is endemic, and during the rainy season it becomes epidemic. Cholera is spread through contaminated water. For several years it has been known that microscopic organisms in the water, called plankton, are the reservoir for the *Vibrio cholerae,* the bacteria causing cholera. Recently, Colwell et al (2003) reported the incidence of cholera was reduced by 48% in Bangladeshi villages using a simple filtering method to treat their drinking water when compared to villages not filtering their water ($P < 0.005$). In this study, 65 villages (comprising over 8,000 households and about 133,000 individuals) were randomly assigned to three groups that used old sari cloth, nylon mesh or nothing to filter their drinking water for an 18-month period[2]. In addition to significantly reducing the cholera incidence, the severity of illness was also less in those villages filtering their water. The researchers suggest this also could be due to the effect of the sari cloth or nylon mesh filters to reduce the number of cholera bacteria in the drinking water.

In this study, sari cloth and nylon mesh were equally effective in preventing cholera and reducing the severity of illness. Old sari cloth is preferred, however, because of its smaller pore size (i.e., filters out plankton and particulates greater than 20 microns), is less expensive and is readily available in Bangladesh as well as many of the countries where cholera is a problem. One could further postulate that by first filtering the contaminated drinking water, followed by treating it with sodium hypochlorite (0.001% final concentration) as described above, an even greater reduction in the incidence of cholera could be obtained at little added cost, especially in rural or remote endemic areas and during epidemics.

REFERENCES

American Dietetic Association (ADA). 1991. *Preparation of Formula for Infants: Guidelines for Health Care Facilities*. ADA: Chicago.

Centers for Diseases Control and Prevention (CDC). 2000. *Safe Water Systems for the Developing World: A Handbook for Implementing Household-Based Water Treatment and Safe Storage Projects.* CDC: Atlanta.

Colwell RR et al. 2003. Reduction of cholera in Bangladeshi villages by simple filtration. *Proc Nalt Acad Sci USA* 100(3): 1051–1055.

Equipment & Systems Engineering (ESE), Inc. 2002. AquaChlor on-site sodium hypochlorite generator. www.aquachlorese.com.

Farr B. 1991. Diarrhea: A neglected nosocomial hazard? *Infect Control Hosp Epidemiol* 12(6): 343–344.

[2] A sari is a traditional garment worn by women in Bangladesh, India and Nepal as well as several other countries in Asia. It is usually made of lightweight cotton that has a thread count of about 140 /inch^2.

Huq A et al. 1996. A simple filtration method to remove plankton-associated *Vibrio cholera* in raw water supplies in developing countries. *Appl Environ Microbiol* 62(7): 2508–2512.

Levine WC et al. 1991. Foodborne disease outbreaks in nursing homes, 1975–1987. *JAMA* 266(15): 2105–2109.

Lynch P et al. 1997. Preventing nosocomial gastrointestinal infections, in *Infection Prevention with Limited Resources*. ETNA Communications: Chicago, pp 125–130.

McFarland LV. 1995. Epidemiology of infectious and iatrogenic nosocomial diarrhea in a cohort of general medicine patients. *Am J Infect Control* 23(5): 295–305.

McFarland LV. 1993. Diarrhea acquired in the hospital. *Gastroenterol Clin North Am* 22(3): 563–577.

McFarland LV et al. 1989. Nosocomial acquisition of *Clostridium difficile* infection. *N Engl J Med* 320(4): 204–208.

Muytjens HL, H Roelofs-Willemse and GH Jaspar. 1988. Quality of powdered substitutes for breast milk with regards to the family Enterobacteriacae. *J Clin Microbiol* 26(4): 743–746.

Villarino ME et al. 1992. Foodbourne disease prevention in health care facilities, in *Hospital Infections*, 3rd ed. Bennett JV and PS Bachman (eds). Little, Brown and Company: Boston, pp 345–358.

TWENTY-SEVEN

PREVENTING PNEUMONIA

KEY CONCEPTS you will learn in this chapter include:

- What type of medical or surgical procedures are most often associated with nosocomial pneumonia
- Importance of cross-contamination (patient to patient or staff to patient) in causing nosocomial pneumonia
- How to minimize the risk of developing nosocomial pneumonia
- How to clean and disinfect respiratory therapy equipment

BACKGROUND

In the US, nosocomial pneumonia is the second most common site of hospital-acquired infections, accounting for 18%. Only nosocomial urinary tract infections are more frequent (Emori and Gaynes 1993). Nosocomial pneumonia also is the infection most likely to be fatal, with mortality rates exceeding 30%, and is the most expensive to treat. Moreover, patients on mechanical ventilators develop pneumonia more frequently and are more likely to have a fatal outcome than those not requiring assisted respiration (Lynch et al 1997). In large part, these findings reflect the severity of the underlying disease.

Most nosocomial pneumonias occur by aspiration of bacteria growing in the back of the throat (oropharynx) or stomach. Intubation and mechanical ventilation greatly increase the risk of infection because they:

- block the normal body defense mechanisms—coughing, sneezing and the gag reflex;
- prevent the washing action of the hair (cilia) and mucus-secreting cells lining the upper respiratory system; and
- provide a direct pathway for microorganisms to get into the lungs.

Other procedures that may increase the risk of infection include oxygen therapy, intermittent positive pressure breathing (IPPB) treatment and endotracheal suctioning.

EPIDEMIOLOGY AND MICROBIOLOGY

Pneumonia is a complex infection that is often difficult to distinguish from other lung diseases, especially adult respiratory distress syndrome, bronchitis, emphysema and congestive heart failure. Most commonly accepted criteria for nosocomial pneumonia include fever, cough,

decreased breath sounds or dullness in a specific area of the lungs and production of purulent (infected) sputum in combination with X-ray evidence suggestive of an infection. If laboratory services are available, typically a gram-stained sputum sample will have many white blood cells (WBCs), bacteria and few epithelial cells but may not be helpful in making the diagnosis. In many countries, additional diagnostic testing (e.g., sputum cultures) is often not available. (Even though specimens from bronchoscopy yield more specific results, bronchoscopy is invasive and the potential complications may outweigh the advantages.)

Half of all nosocomial pneumonias occur after surgery, especially if mechanical ventilation is needed postoperatively. Patients on ventilators, for example, have a 6- to 21-fold greater risk of getting nosocomial pneumonia than do patients not on ventilators (Schaefer et al 1996). While with surgical patients the main reason for mechanical ventilation is the type of operation, for medical patients it usually is related to the patient's illness. Not surprisingly, the risk of postoperative nosocomial bacterial pneumonia is 38 times greater for heart and lung operations (e.g., heart bypass and pulmonary resections) than for surgery at any other site (CDC 1994).

Microbiology

Most reported nosocomial pneumonias are due to bacteria. Early onset pneumonia is likely to involve the patient's own flora, especially streptococcus and haemophilus species. When pneumonia occurs later on during the hospitalization, it is more likely to be due to gram-negative organisms from the hospital environment. The combination of severe illness, presence of multiple invasive devices (IVs, urinary catheters and mechanical ventilators) and frequent contact with the hands of personnel often leads to cross-contamination. For example, in one study by Weinstein (1991), 20–40% of nosocomial pneumonias were due to cross-contamination of organisms from one patient to another, most likely from the hands of hospital staff.

Remember: Handwashing, or use of a waterless, alcohol-based handrub, is an effective way to prevent cross-contamination.

RISK FACTORS

Many risk factors for nosocomial pneumonias are not alterable (e.g., age over 70, chronic lung disease, severe head injuries with loss of consciousness, other serious medical conditions, such as end stage renal disease or cirrhosis). Other risk factors are not alterable during the hospitalization (e.g., cigarette smoking, alcoholism, obesity, major cardiovascular or pulmonary surgery and patients with endotracheal tubes or on ventilators). Although it is impossible to change these risk factors, knowing about them is valuable in terms of anticipating problems and limiting the use of invasive devices (e.g., intravenous lines and urinary catheters) as much a possible. Unfortunately, if the underlying medical or surgical condition is serious, treatment of nosocomial pneumonia may not be successful.

REDUCING THE RISK OF NOSOCOMIAL PNEUMONIA

Preoperative Pulmonary Care

Numerous studies have shown that preoperatively teaching patients about how to prevent postoperative pulmonary problems (e.g., deep breathing, moving in bed, frequent coughing) combined with early movement (sitting up and walking) and limited use of narcotic analgesics for a short duration can reduce the risk of nosocomial pneumonia. The greatest opportunities for prevention of nosocomial pneumonia are in those surgical patients not anticipated to need postoperative ventilation.

Preventing Colonization and Infections with New Organisms

Transfer of organisms among hospitalized patients occurs frequently. Several studies have shown marked reductions in nosocomial colonization and infections when heath workers were required to put on clean gloves (new examination or reprocessed high-level surgical gloves) prior to contact with the mucous membranes and nonintact skin of patients (Lynch et al 1990). Therefore, when caring for patients on mechanical ventilators or receiving IPPB treatment, especially those following heart or lung surgery, it is important to prevent cross-contamination (from staff to patient).

Respiratory Therapy Equipment

To minimize cross-contamination when suctioning patients on ventilators:

- Wash hands or use an alcohol-based antiseptic handrub before putting on gloves.
- Wear clean examination gloves, or reused surgical gloves that have been high-level disinfected, and a protective face shield or mask.
- Remove gloves immediately after therapy is completed and discard them in a plastic bag or leakproof, covered waste container.
- Wash hands or use an alcohol-based antiseptic handrub after removing gloves.

Note: Mechanical ventilation should be used only when necessary and only for as long as necessary.

Suction catheters should be decontaminated, cleaned and high-level disinfected by boiling or steaming between uses. In addition, use of large containers of saline or other fluids for instillation or rinsing the suction catheter should be avoided. If possible, only small containers of sterile solutions or boiled water, which can be used only once and then replaced, should be used.

Remember: Do not touch other items in the room or the patient after suctioning and while still wearing gloves.

To reduce the risk of contamination and possible infection from mechanical respirators and other equipment, the following are suggested:

- Prevent condensed fluid in the ventilator tubing from refluxing into the patient because it contains large numbers of organisms. (Any fluid in the tubing should be drained and discarded, taking care not to allow the fluid to drain toward the patient.)

Remember: Wash hands, or use an antiseptic handrub after handling the tubing.

- Use only small nebulizer bulbs because nebulizers produce aerosols that can penetrate deep into the lungs. (Contaminated large-volume nebulizers have been associated with gram-negative pneumonia and should not be used.)
- Contaminated humidifiers for oxygen administration and ventilator humidifiers are unlikely to cause nosocomial pneumonia because they do not generate aerosols. These humidifiers can, however, be a source of cross-contamination, so they should be cleaned and disinfected between patients.
- Although ventilator circuits may become contaminated at the patient end by organisms from the respiratory tract, there is little evidence that pneumonia is associated with this contamination. Therefore, it is not necessary to change the circuits.

Note: To prevent small volume nebulizer bulbs from becoming contaminated, they should be cleaned and dried between uses, reprocessed daily (decontaminated, cleaned and high-level disinfected by steaming or boiling) and used only with sterile fluids or boiled water.

- Breathing circuits should be decontaminated, cleaned and high-level disinfected by steaming or soaking in a chemical high-level disinfectant.
- Resuscitation devices, such as Ambu bags, are difficult to decontaminate, clean, high-level disinfect and dry between uses. For example, if not thoroughly disinfected and dried, fluids left inside the bag or face piece can be aerosolized during the next use. To prevent this, a good system for prompt reprocessing and return to use is necessary.

Preventing Gastric Reflux

Even short-term (a few days) use of nasal feeding tubes increases the risk of aspiration. Feeding small, frequent amounts rather than large amounts may be less risky. Also, raising the head of the bed, so that the patient is more or less in a sitting position, makes reflux less likely.

Postoperative Management

As mentioned above, surgical patients should be taught how to prevent postoperative pulmonary problems, such as fluid in lungs and/or poorly air-filled areas (atelectasis), preoperatively. Surgical units should have effective plans for:

- optimizing the use of pain medication to keep the patient comfortable enough to cough effectively,
- regularly moving and exercising patients, and
- encouraging deep breathing in the immediate postoperative period and for the next few days.

REFERENCES

Centers for Diseases Control and Prevention (CDC). 1994. Guidelines for prevention of nosocomial pneumonia. Part 1. Issues on prevention of nosocomial pneumonia. Part 2. Recommendations for prevention of nosocomial pneumonia. *Am J Infect Control* 22(4): 247–292. (Authors: Tablan OC et al and HICPAC).

Emori TG and RP Gaynes. 1993. An overview of nosocomial infections, including the role of the microbiology laboratory. *Clin Microbiol Rev* 6(4): 428–442.

Lynch P et al. 1997. Preventing nosocomial pneumonia, in *Infection Prevention with Limited Resources*. ETNA Communications: Chicago, pp 131–134.

Lynch P et al 1990. Implementing and evaluating a system of generic infection precautions: Body substance isolation. *Am J Infect Control* 18(1): 1–12.

Schaefer SD et al. 1996. Respiratory care, in *Pocket Guide to Infection Prevention and Safe Practice*. Mosby-Year Book, Inc.: St. Louis, MO, pp 363–386.

Weinstein RA. 1991. Epidemiology and control of nosocomial infections in adult intensive care units. *Am J Med* 91(3B): 179S–184S.

TWENTY-EIGHT

INFECTION-MONITORING (SURVEILLANCE) ACTIVITIES

KEY CONCEPTS you will learn in this chapter include:

- Why it is important to monitor patient care practices
- What the purpose and limitations of surveillance are
- When surveillance should be considered
- How casefinding can be used to investigate outbreaks of, or exposures to, nosocomial infections
- What some of the common mistakes in investigating outbreaks are

BACKGROUND

Efforts to prevent patients from acquiring an infection or bad outcome (e.g., phlebitis following intravenous infusions) while in a hospital require that healthcare workers use infection prevention practices of demonstrated value and monitor the care being provided. In the broadest sense, infection-monitoring (surveillance) activities are designed to guide **corrective action** based on accurate information, or to provide the rationale for **not acting** when only selective or biased information is available. Poorly designed monitoring activities can, however, waste resources by collecting data that are never used or that fail to provide an accurate picture of what is happening. This occurs most often when surveillance is inconsistent or analysis is incomplete.

Although all healthcare facilities should monitor patient care practices to prevent nosocomial (hospital-acquired) infections and minimize the chance of bad outcomes, surveillance is labor-intensive. Infection surveillance has a long history, and there remains considerable debate about the design, utility and value of surveillance (Lynch et al 1997). How then does a healthcare facility monitor infection-related quality of care activities where resources are limited? As a general rule, monitoring by surveillance should be used only if it will provide specific information not available at less cost. Moreover, it should not consume resources that could be better spent elsewhere. For most facilities with limited resources, the priority should be:

- Ensure recommended infection prevention practices, such as sterilization, or where appropriate HLD, of all items that come in contact with normally sterile tissue, are adhered to.
- Ensure patient care practices are performed according to the best available evidence (i.e., use Standard Precautions for all patients).

- Monitor compliance with recommended practices for certain high-risk procedures, such as inserting central venous catheters.
- Work to eliminate unnecessary and unsafe injections.

Finally, routine surveillance should not outweigh investigating outbreaks, or providing safe water, food and sanitation within the hospital or healthcare facilities. On the other hand, well-organized surveillance ranks ahead of repetitive "staff education" programs, especially those not linked to behavior change activities (**Chapter 3**).

DEFINITIONS

- **Casefinding**. Method of identifying patients with nosocomial infections through a combination of: 1) reviewing medical records, 2) asking questions directed to patients or health workers, and 3) checking laboratory, X-ray or other relevant data, if available.
- **Nosocomial** or **hospital-acquired infection (terms used interchangeably)**. Infection that is neither present nor incubating at the time the patient came to the healthcare facility. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.)
- **Surveillance**. Systematic collection of relevant data on patient care, the orderly analysis of the data and the prompt reporting of the data to those who need it. **Active surveillance** consists of collecting information directly from patients or staff, while **passive surveillance** includes examining reports, laboratory information and data from other sources.

PURPOSE OF SURVEILLANCE

Traditionally surveillance has been used to:

- determine baseline rates of nosocomial infections;
- evaluate infection control measures (e.g., management of multidrug-resistant infections);
- monitor good patient care practices;
- meet the safety standards required by regulatory agencies; and
- detect outbreaks and exposures.

While infection surveillance (collecting some data on all nosocomial infections and calculating rates based on discharges or patient days) is not a useful starting point, knowing when to investigate a situation, what data to collect, how to analyze and interpret the result and how long to measure may be extremely useful. Knowing the difference between monitoring a process (Are they doing what they're supposed to be doing? Why not?)

and monitoring an outcome (Is something bad happening? Who? What? Where? When?) is essential.

Where resources are limited, the use of surveillance as an infection-monitoring tool generally should be restricted to investigating outbreaks or exposures. When considering initiating other types of surveillance activities, the objectives should be reasonable in terms of the resources and time available, and the projected uses for the data should be clearly defined before routine collection of data is established. It is much more difficult to discontinue data collection than to never collect it in the first place.

WHEN TO CONSIDER PERFORMING SURVEILLANCE

Logically, surveillance should begin only after all recommended steps for preventing nosocomial infections have been taken. For hospitals in most countries, rigorously employing the evidence-based infection prevention practices detailed in the preceding **Chapters 3**–**19** should be the primary strategy for preventing nosocomial infections and avoiding bad outcomes in hospitalized patients. Then the use of measures proven to reduce infection risk at specific sites or from invasive procedures should be checked (**Chapters 22**–**27**). Only after successfully implementing and monitoring these recommendations should the use of surveillance be considered.

Finding Patients with Nosocomial Infections

An inexpensive, fairly simple way of finding patients with nosocomial infections is by casefinding. Casefinding consists of reviewing medical records and asking questions of patients and health workers (active surveillance). It is guided by clues obtained from passive surveillance (reports and laboratory information). Routine casefinding is time-consuming and not recommended where resources are limited, but when used to investigate a suspected outbreak (e.g., an increased number of newborns with infectious diarrhea and septicemia over a short time period), casefinding can be extremely helpful.

Using the above example of a suspected outbreak of infectious diarrhea, the **clinical review** of medical records should include collecting basic demographic information (e.g., name, age, date of birth, admission diagnosis), checking for fever, new antibiotic use, new cases of diarrhea, clinical sepsis or the presence of an inflamed surgical wound, drain or IV site. **Talking with patients** (or parents of newborns in this example) should focus on their health, the health of other young children at home, general hygiene, food handling and sanitation. **Discussions with staff** working in the affected area (e.g., the newborn nursery or neonatal ICU) should deal with ensuring that recommended patient care activities (e.g., hand hygiene and use of gloves) are being performed both correctly and at the appropriate times. **Laboratory information** to be checked should include a review of positive cultures and other diagnostic findings if

available. In addition, if laboratory or X-ray staff are informed about the kinds of information that may suggest nosocomial infections, they can alert the infection prevention coordinator or working group with useful tips.

Where time and resources are limited, routine use of casefinding should focus on high-risk areas such as intensive care and postoperative units. In a large study, for example, more than 70% of all nosocomial infections occurred in the 40% of patients who had surgery (Haley et al 1985a and 1985b). Moreover, the infections in these units tended to be more serious than in other areas where infections occur less frequently.

DETECTING AND MANAGING OUTBREAKS

Outbreaks of nosocomial infections do occur, despite the best efforts to prevent them. When they occur, it is important to identify and interrupt the process or practice responsible as quickly as possible to minimize the risk to patients and staff. Investigating and managing suspected outbreaks, however, can be very complex, requiring the assistance of epidemiologists and more experienced infection prevention personnel from national or international health agencies (e.g., CDC). In many instances, however, the cause of the outbreak can be easily identified (i.e., related to a common source, patient care practice or nonpractice) and can be resolved without a complete investigation.

Fortunately, outbreak management is more straightforward, but both require speedy resolution and both are labor and resource intensive. In addition, once the source(s) of the outbreak or exposure is identified, implementing the corrective action may be the most difficult management issue.

Common Mistakes in Outbreak Investigations

Some of the more common errors include:

- Assumption that an outbreak exists when it really does not. An apparent increase in cases over recent experience is often only normal variation; therefore, where possible, confirm the diagnosis, search for additional cases and determine whether the increase is real before concluding that an outbreak is occurring.
- Isolation of an organism rarely explains an outbreak.
- The presence of organisms from multiple sites or personnel usually suggests that these sites became colonized from another source and were not the cause of the outbreak.
- Negative cultures do not justify concluding that the site (e.g., staff or inanimate objects) was not responsible for the outbreak. There could be many reasons the cultures were negative: incorrect specimen collection and handling, poor culture technique, including performing the test incorrectly or using the wrong reagents, and failure to collect the right specimen.

Note: The goal in an outbreak is preventing more patients or staff from becoming infected or at risk.

- Prevention measures are not implemented immediately. As soon as an outbreak is suspected, patient care practices that could be responsible should be evaluated and any problems identified and corrected, without waiting for results from an investigation. (**Table 28-1** outlines common sources for nosocomial infections at various sites and some recommended risk-reduction practices.)
- Other similar practices are not evaluated. When a problem with reprocessing instruments or specific patient care practices is identified, often the same faults exist elsewhere in the hospital; all similar situations should be evaluated and corrected as soon as possible.

Administrative Responsibilities

Hospitalized patients, staff and visitors are all linked to the community at large. In addition, there is considerable interaction between healthcare facilities. Patients may begin care in an ambulance, visit an emergency room, have an inpatient stay and be discharged to a nursing home or receive homecare—all in the same episode of illness. As such, countless health workers, other patients, visitors and staff may be affected. For example, nosocomial outbreaks of measles and hepatitis B have resulted in cases in the community because information regarding an outbreak or exposure in a hospital was not shared. The temptation to withhold this information because it may reflect badly on the hospital, administration or personnel is natural—but must be avoided. Other facilities may have contact with the patients or may use some of the same practices or commercial products that were responsible for the outbreak. Without the frank exchange of information, preventable nosocomial infections may continue to occur. Thus, to minimize the risk to all, the occurrence of exposures and outbreaks should be widely publicized.

Table 28-1. Measures Identified as Effective in Investigating Outbreaks

SITE	WHERE TO LOOK FOR SOURCE AND/ OR MODE		INTERIM MEASURES
	Common	**Uncommon**	
Urinary tract infection	Urinary tract instrumentation Cross-contamination via hands of personnel Poor hand hygiene	Inadequately processed instruments Contaminated antiseptic solution (e.g., povidone-iodine)	Re-emphasize known aseptic practices relating to insertion and maintenance of urinary catheters, and monitor compliance. Institute glove use for any contact with urine. Separate catheterized patients from each other. Put on clean gloves just before contact with urinary meatus. Wash hands, or use an antiseptic handrub, after removal of gloves.
Surgical wounds	Organisms acquired intraoperatively by contact with symptomatic or asymptomatic shedders among staff Contaminated products (wound irrigating solutions) Poor surgical technique (hemostasis, glove puncture)	Airborne spread Preoperative contamination (contaminated antiseptic solution)	Re-emphasize known aseptic practices and surgical technique. Exclude infected personnel from patient care. Separate those at risk from those infected. Put on sterile or high-level disinfected gloves just before wound contact. Use sterile fluids for wound care. Wash hands, or use an antiseptic handrub, after removal of gloves.
Lower respiratory tract	Colonization of upper airway with secondary aspiration into lung Contamination of nebulized solutions or respiratory therapy equipment surfaces Cross-contamination via hands of personnel	Airborne spread	Re-emphasize known aseptic practices and surgical technique. If respiratory therapy is associated with cases, examine technique used for disinfection and delivery of therapies (e.g., multidose vials). Separate those at risk from those infected. Put on clean gloves just before contact with mucous membranes and suctioning of patients. Wash hands, or use an antiseptic handrub, after removal of gloves.
Blood	Intravascular, especially central venous catheters Contamination of insertion site	Inadequately processed instruments Preoperative contamination (contaminated antiseptic solutions)	Re-emphasize known aseptic practices and surgical technique. Intravenous catheters should be changed every 96 hours. Put on sterile or high-level disinfected gloves before inserting catheter and wound contact. Wash hand, or use an antiseptic handrub, after removal of gloves.

Adapted from: Lynch et al 1997.

REFERENCES

Haley RW et al. 1985a. The efficacy of infection surveillance and control programs in preventing nosocomial infections in U.S. hospitals. *Am J Epidemiol* 121(2): 182–205.

Haley RW et al. 1985b. The nationwide nosocomial infection rate: A new need for vital statistics in U.S. hospitals. *Am J Epidemiol* 121(2): 159–167.

Lynch P et al. 1997. Surveillance, outbreak investigations, and exposures, in *Infection Prevention with Limited Resources*. ETNA Communications: Chicago, pp 31–48.

APPENDIX A

GENERAL SURGICAL HANDSCRUB

SUPPLIES

The supplies needed for a surgical handscrub include:

- Plain or antiseptic (preferred) soap (Larson 1988)
- Antiseptic agent (povidone-iodine or chlorhexidine is less irritating to skin)
- Running water (When no running water is available, use a bucket with a tap, which can be turned off to lather hands and turned on again for rinsing, or a bucket and pitcher.)
- Soft stick with pointed end or brush for cleaning underneath the fingernails (These items must be cleaned and preferably high-level disinfected after each use.)
- Soft brush or sponge for cleaning the skin (These items must be cleaned after each use.)[1]
- Towels (sterile towels should be provided in the operating room.)

PROCEDURE

The surgeon, scrub nurse or technician should wear a short-sleeved shirt or blouse when performing a surgical handscrub because it requires scrubbing to the elbows (Sorensen and Luckman 1979).

Procedure	Rationale
1. Remove all jewelry.	1. Jewelry harbors microorganisms, is difficult to clean and makes putting on gloves more difficult (Salisbury 1997).
2. Hold hands above the level of the elbow and wet hands thoroughly. Apply soap, and clean under each fingernail using the stick or brush.	2. Water should flow from area of least contamination (hands) to most contamination (arms). Washing removes many organisms. Fingernails should not extend beyond the tip of the finger more than 3 mm (or 1/8 inch). Long fingernails can puncture gloves, and bacteria grow easily underneath them.

[1] Avoid using stiff scrub brushes as these can damage the skin, especially if surgical handscrub is done several times per day.

Procedure	Rationale
3. Beginning at the fingertips, lather with a soft brush or sponge, using a circular motion. Wash between all fingers. Move from fingertips to the elbow of one arm and repeat for the second arm.	3. Friction and lather raise microorganisms. Moving from area of least contamination to area of most contamination decreases the possibility of spreading contamination.
4. Wash using a soft brush or sponge for at least 2 minutes.	4. If a brush is used, it should be decontaminated and either high-level disinfected or sterilized before reuse; sponges, if used, should be discarded.
5. Rinse each hand and arm separately, fingertips first, holding hands above the level of elbows. Do not let rinse water flow over clean area.	5. Water should flow from area of least contamination to area of most contamination to decrease the possibility of contamination.
6. Apply antiseptic agent and vigorously rub all surfaces of hands, fingers and forearms for at least 2 minutes.	6. Use sufficient antiseptic to cover hands, fingers and forearms.
7. Repeat #5 using clean water.[2]	7. See #5.
8. Use a separate sterile or clean cloth towel for each hand to wipe from the fingertips to the elbow and then discard the towel.	8. Moving from area of least contamination to area of most contamination decreases the possibility of spreading contamination.
9. While waiting to put on sterile or high-level disinfected surgical gloves, hold hands above the level of the waist and do not touch anything.	9. Contact with soiled objects contaminates clean hands. The area below the level of the waist is considered unclean.

Note: If scrubbed hands touch any contaminated surface or object before gloving, Steps 3 through 9 must be repeated.

REFERENCES

Colwell RR et al. 2003. Reduction of cholera in Bangladeshi villages by simple filtration. *Proc Nat Acad Sci USA* 100(3): 1051–1055.

Larson EL. 1988. Guideline for use of topical antimicrobial agents. *Amer J Infect Control* 16(6): 253–266.

Salisbury DM et al. 1997. The effect of rings on microbial load of health care workers' hands. *American Journal of Infection Control* 25(1): 24–27.

Sorensen KC and J Luckman. 1979. *Basic Nursing: A Psychophysiologic Approach*. WB Saunders Co.: Philadelphia, pp 934–938.

Huq A et al. 1996. A simple filtration method to remove plankton-associated *Vibrio cholera* in raw waters supplies in developing countries. *Appl Environ Microbiol* 62(7): 2508–2512.

[2] If tap water is cloudy, most particulates (debris and organic material) can be removed by filtering through four layers of moderately woven cotton cloth, such as cheese cloth or old sari material, before boiling or treating with dilute chlorine (sodium hypochlorite) solution (Colwell et al 2003; Huq et al 1996).

APPENDIX B

ANTISEPTICS

Many chemicals qualify as antiseptics. The following antiseptics are generally available in many parts of the world:

- 60–90% alcohols (ethyl, isopropyl or "methylated spirit")
- 2–4% chlorhexidine gluconate (Hibiclens®, Hibiscrub®, Hibitane®)
- Chlorhexidine gluconate and cetrimide, various concentrations (Savlon®)
- 3% iodine; aqueous iodine and alcohol-containing (tincture of iodine) products
- 7.5–10% iodophors, various concentrations (Betadine® or Wescodyne®)
- 0.5–4% chloroxylenol (Para-chloro-metaxylenol or PCMX) various concentrations (Dettol®)
- 0.2–2% triclosan

In choosing an antiseptic, the desired characteristics (e.g., absorption and persistence) should be considered along with evidence of a given product's safety and efficacy, its acceptability to staff and, most importantly, its cost (Boyce and Pittet 2002; Larson 1995; Rutala 1996). **Table B-1** lists several recommended antiseptic solutions, their microbiologic activity and potential uses. (The grading system used in this table is excellent, good, fair, and none.)

ALCOHOL SOLUTIONS (ETHYL OR ISOPROPYL)

Note: In many countries, alcohols are available as "industrial methylated spirit," or ethyl alcohol denatured with a small amount of wood (methyl) alcohol (Harpin and Rutter 1982). Because methyl alcohol is the least effective of the alcohols, it should not be used alone as an antiseptic or disinfectant. Before using, be sure the ethyl alcohol is of adequate strength (60–90%) in locally available "spirit."

Ethyl and isopropyl alcohol (60–90%) are excellent antiseptics that are commonly available and inexpensive. Their rapid killing action makes them very effective in reducing numbers of microorganisms on skin, even under gloves. Alcohols are effective against all hepatitis viruses and HIV. They should **not** be used on mucous membranes (e.g., for vaginal preparation). (Alcohols dry and irritate mucous membranes which, in turn, promotes the growth of microorganisms.)

Alcohols are among the safest known antiseptics. A 60–70% solution of ethyl or isopropyl alcohol is effective, less drying to the skin and less expensive than higher concentrations. Because it is less drying to the skin, ethyl alcohol may be more appropriate than isopropyl alcohol for frequent use on skin (Larson 1995).

Table B-1. Antiseptics: Microbiologic Activities and Potential Uses

GROUP	ACTIVITY AGAINST BACTERIA							POTENTIAL USES			
	Gram-Positive	**Most Gram-Negative**	**TB**	**Viruses**	**Fungi**	**Endospores**	**Relative Speed of Action**	**Affected by Organic Matter**	**Surgical Scrub**	**Skin Preparation**	**Comments**
Alcohols (60–90% ethyl or isopropyl)	Excellent	Excellent	Excellent	Excellent	Excellent	None	Fast	Moderate	Yes	Yes	Not for use on mucous membranes Not good for physical cleaning of skin, no persistent activity
Chlorhexidine (2–4%) (Hibitane, Hibiscrub)	Excellent	Good	Fair	Excellent	Fair	None	Intermediate	Slight	Yes	Yes	Has good persistent effect Toxicity to ears and eyes
Iodine preparations (3%)	Excellent	Excellent	Excellent	Excellent	Good	Fair	Intermediate	Marked	No	Yes	Not for use on mucous membranes Can burn skin so remove after several minutes
Iodophors (7.5–10%) (Betadine)	Excellent	Excellent	Fair	Good	Good	None	Intermediate	Moderate	Yes	Yes	Can be used on mucous membranes
Para-chloro-metaxylenol (PCMX) (0.5–4%)	Good	Excellent	Fair	Good	Fair	Unknown	Slow	Minimal	No	Yes	Penetrates the skin and should not be used on newborns
Triclosan (0.2–2%)	Excellent	Good	Fair	Excellent	None	Unknown	Intermediate	Minimal	Yes	No	Acceptability on hands varies

Adapted from: Olmsted 1996; Boyce and Pittet 2002.

Advantages

- Rapidly kill all fungi and bacteria including mycobacteria; isopropyl alcohol kills most viruses, including HBV, HCV and HIV; ethyl alcohol kills all viruses.
- Although alcohols have no persistent killing effect, the rapid reduction of microorganisms on skin protects against regrowth of organisms, even under gloves, for several hours.
- Are relatively inexpensive and widely available throughout the world.

Disadvantages

- Need emollient (e.g., glycerin or propylene glycol) to prevent drying of skin (ethyl alcohol may be less drying than isopropyl).
- Easily inactivated by organic materials.
- Flammable (requires storage in cool, well-ventilated areas).[1]
- Will damage rubber (latex) over time.
- Not good cleaning agents.

CHLORHEXIDINE GLUCONATE (CHG)

Chlorhexidine gluconate (CHG) is an excellent antiseptic. It remains active against microorganisms on skin many hours after use (residual effect) and is safe even for use on newborn infants. Because CHG is inactivated by soap, its residual antimicrobial activity is dependent upon the concentration of CHG in the commercial product. Two to four percent chlorhexidine is the recommended concentration. New 2% aqueous formulations and 1% chlorhexidine in a waterless, alcohol-based antiseptic handrub also are effective (Larson 1995).

Advantages

- Broad spectrum of antimicrobial action.
- Persistent action on skin (chemically active for at least 6 hours).[2]
- Chemical protection (the number of microorganisms inhibited) increases with repeated use.
- Minimally affected by organic material.
- Several products available commercially, most common is in detergent base or as a waterless, alcohol-based antiseptic handrub.

Disadvantages

- Expensive and not always available.
- Action reduced or neutralized by natural soaps, by substances present in hard tap water and some hand creams.
- Not effective against tubercle bacillus, only fairly active against fungi.

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[1] Residual alcohol on hands or skin may be ignited by static electricity, so allow hands to dry thoroughly after using antiseptic handrub.

[2] For maximum effectiveness and residual activity, chlorhexidine should be used on a regular basis (at least daily).

- Cannot be used above pH of 8 because it decomposes.
- Avoid contact with eyes as it can cause conjunctivitis.

IODINE AND IODOPHOR SOLUTIONS

Three percent iodine solutions are very effective antiseptics and are available as both aqueous (Lugol) and tincture (iodine in 70% alcohol) solutions. Seven and a half percent to ten percent iodophors are solutions of iodine mixed with a carrier, a complexing agent such as polyvinyl pyrrolidone (povidone) that releases small amounts of iodine. Povidone-iodine is the most common iodophor and is available globally.

Note: Iodophors manufactured for use as antiseptics are **not** effective for disinfecting inorganic objects and surfaces. These iodine solutions have significantly less iodine than chemical disinfectants (Rutala 1996).

The amount of "free" iodine present determines the level of antimicrobial activity of iodophors (e.g., 10% povidone-iodine contains 1% available iodine, yielding a "free" iodine concentration of 1 ppm [0.0001%]) (Anderson 1989). Iodophors have a broad spectrum of activity. They kill vegetative bacteria, mycobacterium, viruses and fungi; however, they **require up to 2 minutes of contact time to release free iodine**, which is the active chemical. Once released the free iodine has rapid killing action. In addition, iodophors generally are nontoxic and nonirritating to skin and mucous membranes unless the person is allergic to iodine (Larson 1995).

Advantages

- Broad spectrum of antimicrobial action.
- Aqueous iodine preparations are inexpensive, effective and widely available.
- Iodophors are nonirritating to skin or mucous membranes (unless the person is allergic to iodine), making them ideal for vaginal use (e.g., before IUD insertion).
- Up to 3% aqueous solutions do not stain skin.

Disadvantages

- Slow to intermediate antimicrobial action.
- Iodophors have little residual effect.
- Like alcohols, iodine and iodophors are rapidly inactivated by organic materials, such as blood or sputum.
- Tincture or aqueous iodine may cause skin irritation and staining, and must be removed from skin after drying. (Use alcohol to remove the stain.)
- Absorption of free iodine through skin and mucous membranes may cause hypothyroidism in newborn infants so use should be limited (Newman 1989).
- Allergic reactions to iodine and iodophors can occur, so check patient for history of allergy (iodine and shellfish).

Note: Iodine (aqueous or tincture) must never be used on mucous membranes because of its rapid absorption and irritation to the epithelium.

Do not dilute commercially available iodophors manufactured for antisepsis (Betadine or Wescodyne) as this increases the concentration of "free" iodine that can be released and increase the degree of skin irritation.

CHLOROHEXYLENOL

Note: In commercial preparations such as Dettol, which is expensive, the antiseptic and disinfectant activity is due primarily to the alcohol content, not the chlorohexylenol. A 60–90% alcohol solution is equally effective and much less expensive.

Chlorohexylenol (para-chloro-metaxylenol or PCMX) is a halogenated derivative of xylenol that is widely available in concentrations of 0.5–4%. Chlorohexylenol destroys microorganisms by breaking down the cell wall. It has low germicidal activity (Favero 1985) compared to alcohols, iodine and iodophors and is less effective in decreasing skin flora than either CHG or iodophors (Sheena and Stiles 1982). Because it penetrates the skin, it may be toxic when applied to some areas of the body, and should not be used on newborns. Therefore, commercial products with chlorohexylenol concentrations above 4% should not be used.

Advantages

- Broad spectrum of activity.
- Only minimally affected by organic materials.
- Residual effect persists for several hours.
- Minimally affected by organic matter.

Disadvantages

- Inactivated by soaps (nonionic surfactants), making it less useful for skin preparation.
- Should not be used on newborns due to rapid absorption and potential toxicity.

TRICLOSAN

Triclosan is a colorless substance that has been incorporated into soaps as an antimicrobial agent. Concentrations from 0.2–2.0% have moderate antimicrobial activity against gram-positive cocci, mycobateria and yeast but not gram-negative bacilli, especially *P. aeruginosa* (Larson 1995). Although concern has been expressed that resistance to this agent may develop more readily than with other antiseptic agents, resistance to skin flora has not been observed in long-term clinical studies to date.

Advantages

- Broad spectrum of activity.
- Excellent persistence.
- Minimally affected by organic matter.

Disadvantages

- Not affective against *P. aeruginosa* or other gram-negative bacilli.
- Bacteriostatic (only inhibits growth).

PRODUCTS THAT SHOULD NOT BE USED AS ANTISEPTICS

Hexachlorophene (HCP)

Three percent hexachlorophene is active against gram-positive cocci such as staphylococcus, but has little or no activity against gram-negative bacteria, viruses, *Mycobacterium tuberculosis* and fungi. It is slow acting, and one wash with hexachlorophene does not reduce skin flora. Hexachlorophene has neurotoxic side effects and can penetrate the skin of premature infants. It should never be used on broken skin or mucous membranes. Also, when it is used intermittently, bacteria may grow back in large numbers between uses (rebound growth), all of which limits use (Larson 1995).

Zephiran® (benzalkonium chloride)

Zephiran is commonly used in many parts of the world as an antiseptic, but it has several distinct disadvantages:

- Solutions of benzalkonium chloride have repeatedly been shown to become contaminated by *Pseudomonas* species and other common bacteria (Block 1991).
- Solutions of benzalkonium chloride are easily inactivated by cotton gauze and other organic material and are incompatible with soap (Block 1991).
- Zephiran takes at least 10 minutes to kill HIV, the virus causing AIDS (Angle 1992). By contrast, 0.5% chlorine solution kills HIV in less than 1 minute.

Mercury Laurel or Other Mercury-Containing Compounds

Although frequently sold as antiseptics, chemicals containing mercury should be avoided because of their **high toxicity** (Block 1991):

- Skin exposure to low levels of mercury causes blister formation and contact dermatitis.
- Inhalation or ingestion of low levels of mercury affects the central nervous system (numbness, speech impairment, deafness), and higher levels (200 mg) are fatal.
- Skin contact alone can result in absorption of measurable amounts of mercury.
- Pregnant women exposed to small doses may not show toxic effects themselves. The fetus, however, may be harmed because mercury is a **potent** teratogen (causes birth defects, including cleft palate, cerebral palsy and other central nervous system abnormalities).

REFERENCES

Anderson RI. 1989. Iodophor antiseptics: Intrinsic microbial contamination with resistant bacteria. *Infect Control Hosp Epidemiol* 10: 443–446.

Angle M. 1992. Infection Prevention in FP/MCH, in *Guidelines for Clinical Procedures in Family Planning*. INTRAH: Chapel Hill, NC.

Block SS. 1991. *Disinfection, Sterilization and Preservation*, 4th ed. Lea & Febiger: Philadelphia.

Boyce JM and D Pittet. 2002. Guidelines for hand hygiene in healthcare settings: Recommendations of the Healthcare Infection Control Practices Advisory Committee and the HICPAC/SHSA/APIC/IDSA Hand Hygiene Task Force. *Infect Control Hosp Epidemiol* 23(Suppl): S3–S40. Available at: http://www.cdc.gov/handhygiene.

Favero MS. 1985. Sterilization, disinfection, and antisepsis in the hospital, in *Manual of Clinical Microbiology,* 4th ed. American Society for Clinical Microbiology: Washington, DC, pp 129–137.

Harpin V and N Rutter. 1982. Percutaneous alcohol absorption and skin necrosis in a preterm infant. *Arch Dis Child* 57(6): 478.

Larson EL. 1995. APIC guideline for handwashing and hand antisepsis in health care settings. *Amer J Infect Control* 23(4): 251–269.

Newman NM. 1989. Use of povidone-iodine in umbilical cord care. *Clin Pediatrics* 28(1): 37.

Olmsted RN (ed). 1996. *Infection Control and Applied Epidemiology: Principles and Practices*. Association for Practitioners in Infection Control (APIC), Table 19-2. CV Mosby: St. Louis, MO.

Rutala WA. 1996. APIC guideline for selection and use of disinfectants. *Amer J Infect Control* 24(4): 313–342.

Sheena AZ and ME Stiles. 1982. Efficacy of germicidal handwashing agents in hygienic hand disinfection. *Br J Med* 65: 855–858.

APPENDIX C

PROCESSING SURGICAL GLOVES

The risk in reusing surgical gloves is that processed gloves have more inapparent tears than new ones and therefore provide less protection to the wearer. Sterilization (autoclaving) and high-level disinfection (steaming) of gloves, when correctly performed, however, can provide a high quality product (**Chapter 14**). In addition, **double gloving** for high-risk procedures can be done. Therefore, processing surgical gloves constitutes an **appropriate reuse of disposable items** where resources are limited (Daschner 1993).

HOW TO DECONTAMINATE AND CLEAN SURGICAL GLOVES BEFORE STERILIZATION OR HIGH-LEVEL DISINFECTION (HLD)

STEP 1: Before removing soiled surgical gloves, immerse hands briefly in a container filled with 0.5% chlorine solution.

STEP 2: Remove gloves by turning inside out and soak them in the chlorine solution for 10 minutes.

(Performing **Steps 1 and 2** insures that both surfaces of the gloves are decontaminated.)

Note: Latex rubber surgical gloves should be discarded after processing three times because the gloves tear more easily with additional processing (Bagg, Jenkins and Barker 1990; Martin et al 1988).

STEP 3: Wash gloves in soapy water, cleaning inside and out.

STEP 4: Rinse gloves in clean water until no soap or detergent remains. (Residual soap or detergent can interfere with sterilization or HLD.)

STEP 5: Test gloves for holes by inflating them by hand and holding them under water. (Air bubbles will appear if there are holes.)

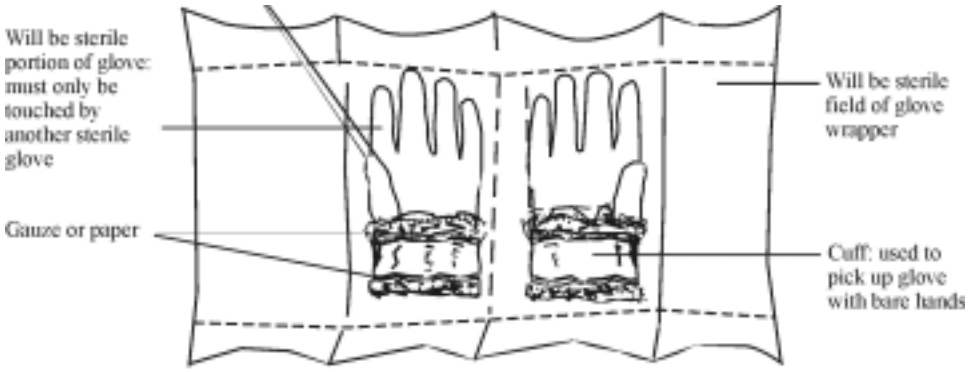
STEP 6: Gently air dry gloves inside and out before proceeding with sterilization. (Gloves which remain wet for long periods of time will absorb water and become tacky.)

HOW TO STERILIZE SURGICAL GLOVES

Remember: Higher temperatures and pressures are destructive to gloves.

After decontamination, cleaning and drying, gloves must be packaged prior to sterilizing by autoclaving. First, fold the cuffs of the gloves out toward the palm so that after sterilization they can be put on easily and without contamination. Next, put gauze or paper inside each glove and under the fold of the cuff and wrap the gloves as shown in **Figure C-1**. (Do not tie tightly or wrap glove packs with rubber bands.) Finally, place them in a wire basket on their sides to allow optimum steam penetration. (If gloves are stacked in piles, penetration of steam under the cuffs may be poor.) Autoclave at 121°C (250°F) for 30 minutes and at a pressure of 106 kPa (15 lb/in^2).

Figure C-1. Preparing Gloves for Autoclaving (Steam Sterilization)



Source: South East Asia Office (SEARO)/ World Health Organization 1988.

Immediately after autoclaving, gloves are extremely friable and tear easily. Gloves should **not** be used for 24 to 48 hours to allow their elasticity to return and to prevent tackiness (stickiness) (**Table C-1**).

Table C-1. Tips to Help Avoid Glove Problems

Probable Cause	Recommended Solution
PROBLEM: TACKY OR STICKY GLOVES	
Residual liquid soap or detergent	Reduce amount of liquid soap or detergent used when washing gloves.
	Rinse gloves at least three times in clean water.
Heated to high temperature for too long	Use **30 minutes** sterilizing time at 121°C (250°F) and remove gloves from sterilizer as soon as cycle is completed.
Gloves sterilized with other goods	Sterilize gloves separately.
Gloves not allowed to dry completely after steaming	Wear "wet" within 30 minutes or allow to dry for 4 to 6 hours before using.
Surfaces of gloves touching each other	Gauze or paper wicks should be inserted between the palm and back of hand of each glove and between the hand of the glove and the turned-back cuff. This allows steam to contact all surfaces during sterilization and prevents surfaces from adhering to each other.
Breakdown (deterioration) of rubber (latex)	Store in a dry, cool area.
(Rubber gloves deteriorate while stored even though they have not been used. They become soft, sticky and unusable.)	Do not store in direct sunlight.
PROBLEM: EXCESSIVE TEARING OR RUPTURING	
Gloves used too soon following sterilization	**Do not** use gloves for 24 to 48 hours after sterilization. This allows gloves to regain their elasticity before use.

Source: Tomlinson 1991.

HOW TO HIGH-LEVEL DISINFECT SURGICAL GLOVES BY STEAMING

After surgical gloves have been decontaminated and thoroughly washed, they are ready for HLD by steaming (McIntosh et al 1994). (See **Chapter 12** for more information on steaming.)

STEP 1: Fold up the cuffs of the gloves so that they can be put on easily and without contamination after HLD.

STEP 2: Place gloves into one of the steamer pans that has holes in its bottom. To make removal from the pan easier, the cuffs should be facing outward toward the edge of the pan (**Figure C-2**). Five to fifteen pairs can be put in each pan depending on the size (diameter) of the pans.

Figure C-2. Gloves in Steamer Pan

STEP 3: Repeat this process until up to three steamer pans have been filled with gloves. Stack the filled steamer pans on top of a bottom pan containing water for boiling. A second empty, dry bottom pan (without holes) should be placed on the counter next to the heat source (see **Step 9**).

STEP 4: Place the lid on the top pan and bring water to a full **rolling** boil. (When water only simmers, very little steam is formed and the temperature may not get high enough to kill microorganisms.)

Remember: Be sure there is sufficient water in the bottom pan for the **entire** 20 minutes of steaming.

STEP 5: When steam begins to come out between the pans and the lid, start the timer or note the time on a clock and record the time in the HLD log.

STEP 6: Steam gloves for 20 minutes.

STEP 7: Remove the top steamer pan and put the lid on the pan that was below it (the pan now on top). Gently shake excess water from the pan just removed.

STEP 8: Place pan just removed onto the empty bottom pan (see **Step 3**). Repeat until all pans containing gloves are restacked on this empty pan and the top pan is covered with the lid. (This step allows the gloves to cool and dry without becoming contaminated.)

Remember: Do **not** place pans containing gloves on a table top, counter or other surface as gloves will be contaminated.

STEP 9: Allow gloves to air dry in the steamer pans (4 to 6 hours) before using.[1]

STEP 10: Using a high-level disinfected forceps, transfer the dry items to a dry, high-level disinfected container[2] with a tight-fitting cover. Gloves can also be stored in the stacked and covered steamer pans as long as a bottom pan (no holes) is used.

REFERENCES

Bagg J, S Jenkins and GR Barker. 1990. A laboratory assessment of the antimicrobial effectiveness of glove washing and re-use in dental practice. *J Hosp Infect* 15(1): 73–82.

Daschner F. 1993. The hospital and pollution: Role of the hospital epidemiologist in protecting the environment, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RP (ed). Williams & Wilkins: Baltimore, MD.

Martin MV et al. 1988. A physical and microbial evaluation of the re-use of non-sterile gloves. *Br Dent J* 165(9): 321–324.

McIntosh N et al. 1994. *Practical Methods for High-Level Disinfection of Surgical Gloves.* Paper presented at American Public Health Association Annual Meeting. Session no. 2285, Washington, D.C., 31 October–4 November.

South East Asia Office (SEARO)/World Health Organization (WHO). 1988. *Manual on Infection Control in Health Facilities*. SEARO Regional Health Papers No. 18: New Delhi.

Tomlinson M. 1991. Personal reference. Chosen Mission Project: Erie, PA.

[1] Alternatively, allow gloves to cool for 5 to 10 minutes before wearing "wet." Gloves should be used within 30 minutes, if possible. After this time, the fingers of the gloves stick together, and the gloves are hard to put on despite being damp. Gloves that have been removed from the steamer pan(s) to be worn "wet" but were not used during the clinic session should be reprocessed before using.

[2] How to prepare a high-level disinfected container: For small containers, boil water in the covered container for 20 minutes, then pour out the water, which can be used for other purposes, replace the cover and allow container to dry. Alternatively, and for large containers, fill a plastic container with 0.5% chlorine solution and immerse the cover in chlorine solution as well. Soak both for 20 minutes. (The chlorine solution can then be transferred to another container and reused.) Rinse the cover and the inside of the container three times with boiled water and allow to air dry.

APPENDIX D

PRECAUTIONS FOR THE SURGICAL TEAM

Safety in the operating room, both for patients and staff, requires careful planning, use of appropriate personal protective equipment (PPE) and demands daily attention and maintenance by the surgical team members and support staff. While traditionally the focus of attention in the OR has been almost totally directed to protecting the patient, the emergence of the HIV/AIDS crisis, increasing HCV rates and resurgence of tuberculosis necessitate that equal attention be given to protecting health workers and professional staff. In this new era, each member of the team must develop the habit of focusing on both patient safety and occupational safety at the same time.

The following section contains safety checklists for the surgical team that have been adapted from an operating room safety manual by Davis (2001). They are intended to serve as general guides to improving safety in the operating room. In addition, they serve as reminders and as means of raising awareness of risk. These checklists are "not set in stone." They should be tailored to procedures and personnel, regularly reviewed and updated as new knowledge and safety practices evolve.

GENERAL SAFETY CHECKLIST (FOR SURGICAL TEAM)

Absolute prerequisites

- ❑ Complete the hepatitis B vaccination series.
- ❑ Use Standard Precautions with all patients.

Personal protective equipment—appropriate choices

- ❑ Wear fluid-resistant head wear where appropriate.
- ❑ Use adequate eye and face protection.
- ❑ Use appropriate neck protection (consider recently shaved skin as nonintact).
- ❑ Wear fluid-resistant or fluid-impervious gowns, as appropriate to expected exposure risk (if available).
- ❑ Choose gloves appropriately (use double gloving, see below).
- ❑ Wear appropriate footwear (shoes not open toed or flip flops).

Personal protective equipment—appropriate use

- ❑ Remove gloves carefully to avoid blood splatter.
- ❑ Wash hands with soap and clean water or use antiseptic handscrub after removing gloves.
- ❑ Remove eye protection last.
- ❑ Remove contaminated personal protective equipment (PPE) before leaving the room.
- ❑ Carefully remove and discard mask following every procedure.

Safety techniques

- ❑ Wear examination gloves when handling surgical specimens.
- ❑ Wear eye protection if container is opened or splashing is anticipated.
- ❑ Apply dressings and handle drains or packs wearing clean new examination gloves.
- ❑ Avoid touching any surface with contaminated gloves.

Safety strategies

- ❑ Have extra PPE readily available should replacements be needed.
- ❑ Position sharps disposal containers at point of use.
- ❑ Have a plan for sharps management.
- ❑ Make sure all team members know the plan.
- ❑ Modify the plan as needed.
- ❑ Focus attention on sharps in use; be aware and alert.
- ❑ Alert other OR team members to possible hazards.
- ❑ Discourage unauthorized entry into the room.
- ❑ Keep extraneous conversation to a minimum.
- ❑ Store a tube of blood preoperatively on all surgical patients to be held in the laboratory for possible HIV testing should an exposure occur.
- ❑ A signed consent for HIV testing, in case of an exposure, should be obtained preoperatively to avoid delay in post-exposure followup.

Personal preparation

- ❑ Prepare your body and mind to function effectively and efficiently.
- ❑ Get enough sleep before surgery. If you are working a long shift on obstetrics or trauma service, nap if and when you can.
- ❑ Avoid caffeine, which increases hand tremor.
- ❑ Avoid alcohol or other substances that impair perception, judgment or reflexes.
- ❑ Promote general good health. Exercise regularly and have an annual physical.
- ❑ Avoid behaviors that increase nonoccupational risk of exposure to bloodborne viruses, such as unsafe sex.

SAFE ASSISTING AND OPERATING CHECKLIST

- ❑ Use forceps to put scalpel blade on handle.
- ❑ Avoid handling suture needles manually.
- ❑ Never hold a scalpel, loaded needle holder, or any other sharp in the same hand simultaneously with another instrument.
- ❑ Scalpels, loaded needle holders, and other sharps should be held in the hand only during cutting, suturing, or for other specific tasks. At all other times, sharps should be placed off the operative field.
- ❑ Properly employ a Safe Zone for the safe passing of sharps.
- ❑ Use verbal warnings to announce transfer of sharps.
- ❑ Before tying, either remove the needle from the suture and park the needle safely, or protect the needle point with the needle holder.
- ❑ Avoid finger contact with tissue being sutured or cut.
- ❑ Use retractors rather than manual retracting whenever possible.
- ❑ Avoid reflex sponging of tissue, which may not be anticipated by the surgeon, when a sharp is in use.
- ❑ Keep eyes on all sharps in use until they are returned to the Safe Zone.
- ❑ Pass long laparoscopic instruments, such as needle tip cautery and sharp-pointed scissors, handle first and tip down.
- ❑ Replace the shield onto the tip of a drain trocar with an instrument, not the fingers, before pulling the trocar out of the exit wound.
- ❑ When doing repeat injections with hypodermic needle and syringe, stick needle in rolled, sterile towel when not in use.
- ❑ Remove scalpel blade using forceps; place in sharps container.

OPERATING ROOM SAFETY CHECKLIST
(Post at scrub sinks or at OR doors)

PERSONAL PROTECTIVE EQUIPMENT

- ❑ Head wear that covers scalp hair
- ❑ Eye and face protection in place
- ❑ Appropriate gown
- ❑ Impervious boots
- ❑ Double gloves or glove liners as indicated
- ❑ Waterproof drapes (if available)

WORK PRACTICE CONTROLS

- ❑ Safe Zone selected and deployed
- ❑ Surgeon( ) right-handed ( ) left-handed
- ❑ Assistant ( ) right-handed ( ) left-handed
- ❑ Appropriate suture needle selection (blunt if applicable)
- ❑ Appropriate retractor selection (blunt if applicable)
- ❑ Disposable scalpels (if available)
- ❑ Smoke evacuation equipment available and functioning (operative laparoscopy)

SHARPS MANAGEMENT

- ❑ Sharps disposal container

DELIVERY ROOM SAFETY CHECKLIST
(Post at scrub sinks or at OR doors)

PERSONAL PROTECTIVE EQUIPMENT

- ❑ Head wear that covers scalp hair
- ❑ Eye and face protection in place
- ❑ Appropriate gown
- ❑ Impervious boots/shoe covers
- ❑ Double gloves, extended cuff gloves (gauntlet) or glove liners as indicated
- ❑ Waterproof drapes (if available)

WORK PRACTICE CONTROLS

- ❑ SAFE Zone (cesarean section)
- ❑ Surgeon ( ) right-handed ( ) left-handed
- ❑ Assistant ( ) right-handed ( ) left-handed
- ❑ Appropriate suture needle selection (blunt if applicable for episiotomy)
- ❑ Appropriate retractor selection (blunt if applicable for cesarean section)

SHARPS MANAGEMENT

- ❑ Sharps disposal container

MINIMALLY INVASIVE SURGERY SAFETY CHECKLIST

- ❑ Pass trocars, needles, and other short-length sharps through a Safe Zone.
- ❑ Pass long laparoscopic instruments that don't fit in the Safe Zone, such as needle-tip cautery and sharp-pointed scissors, handle first and tip down.
- ❑ Place long-pointed cautery needles, hollow-bore needles or other long sharps into sleeve ports, on request only, using two hands—preferably one person's hands—and then angle the handle toward the surgeon's waiting hand.
- ❑ Blunt-tipped suture needles may be used effectively during laparoscopic hysterectomy and are considered a safer option for patient and surgeon.
- ❑ Avoid sprayback; use trocar valves to protect anesthesia personnel as well as the surgical team members.
- ❑ Aspirate all gas, fluid, and blood from the abdomen prior to closing.

SAFE SHARPS DISPOSAL CHECKLIST

- ❑ Choose containers with built-in safety features, such as "see-through" (translucent) boxes with a readily apparent three fourths and full level lines.
- ❑ Lids should allow the sharp to enter the container by gravity alone, without the need for additional manipulation.
- ❑ Install containers close to the point of use ideally within arm's reach.
- ❑ Mount containers at a convenient height for use and service, in plain sight and free from obstructions.
- ❑ Do not leave containers freestanding on the floor on their side.
- ❑ Do not shake containers to avoid spillage or sharps sticking out.
- ❑ Schedule staff training and education for proper use of sharps containers.
- ❑ Assign responsibility for maintenance and service of sharps containers.

REFERENCES

Davis MS. 2001. *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures*, 2nd ed. Sweinbinder Publications LLC: Atlanta.

APPENDIX E

DECONTAMINATING AND CLEANING SURGICAL INSTRUMENTS AND HYPODERMIC NEEDLES AND SYRINGES

HOW TO DECONTAMINATE AND CLEAN SURGICAL (METAL) INSTRUMENTS

Decontamination

STEP 1: After use, immerse all instruments in a plastic container filled with 0.5% chlorine solution or other locally available disinfectant for 10 minutes for decontamination. (This step is necessary to help prevent transmission of HBV or HIV/AIDS to clinic staff.)

STEP 2: If the instruments and other items cannot be cleaned immediately, rinse the objects with water and towel dry to minimize possible corrosion

Cleaning

STEP 3: Scrub instruments under the surface of the water to prevent splashing of infectious materials. Use a soft brush and liquid soap or detergent and water. Be sure to clean the teeth, joints and screws—an old toothbrush works well.

Remember: When cleaning instruments and other items, wear utility gloves and, if available, protective eyewear, a facemask and a plastic or rubber apron.

Do not use hot water because it coagulates protein, making blood and body fluids hard to remove.

STEP 4: Rinse with clean water until no soap or detergent remains.[1] Soap or detergent can interfere with the action of some chemical disinfectants.

STEP 5: Dry by air or with a clean towel. Water from wet instruments will dilute chemicals used for sterilization or high-level disinfection (HLD), making them ineffective. (Drying is not necessary for instruments that are to be high-level disinfected by boiling or steaming.)

STEP 6: Proceed with sterilization (if available) or HLD (see **Chapter 11** or **12**).

HOW TO DECONTAMINATE, DISPOSE OF OR REPROCESS HYPODERMIC NEEDLES AND SYRINGES

The processing and disposal of **hypodermic needles** and **syringes** constitute a special problem. Clearly, to minimize the risk of needlestick injuries to staff and because needles are difficult to clean and either sterilize or high-level disinfect satisfactorily, they should be reused as little as possible.

[1] If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001% (see **Chapter 26**).

Processing used needles represents an **inappropriate reuse of disposables** and is responsible for numerous infections (Kane et al 1999; Phillips et al 1971; Simonsen et al 1999). In some circumstances (where resources are limited), however, it is the only available option.

Instructions

When available and affordable, single-use **disposable** sterile plastic syringes and needles or one of the new autodisable syringes are recommended for all patient care use. If disposables are being used, it is important to:

- Maintain adequate supplies.
- Decontaminate assembled needles and syringes and discard them in a puncture-resistant sharps container immediately after use.[2]
- Dispose of these containers by burning, encapsulating or burying them.

As mentioned above, if needles and syringes will be reused, the safest approach is to process **only** syringes, but **not** needles. For those situations where **both needles** and **syringes** must be reused, care must be taken to avoid accidental needlesticks to cleaning staff during processing. Instructions for disposing of the needle and syringe or reprocessing either the syringe alone or both items are given below.

Disposal of Needle and Syringe

STEP 1: Do not recap needle or disassemble needle and syringe.

STEP 2: After use, to decontaminate the needle and syringe, hold the needle tip under the surface of a 0.5% chlorine solution, fill with solution and push out (flush) three times.

Note: Sharps containers should be placed close to the area they will be used—within arm's reach if possible.

STEP 3: Place the assembled needle and syringe in a puncture-resistant sharps container such as a heavy cardboard box, plastic bottle or tin can with lid.

STEP 4: When the container is three-quarters full, seal and either burn, encapsulate or bury it.

Disposal of Needle but Syringe Reused

STEP 1: Do not recap needle or disassemble needle and syringe.

STEP 2: Immediately after use, fill the syringe with a 0.5% chlorine solution by drawing it into the syringe through the needle.

STEP 3: Decontaminate assembled needle and syringe by placing in a 0.5% chlorine solution for 10 minutes.

STEP 4: Wearing utility gloves, remove the needle and syringe from decontamination solution, and push out (flush) solution from the assembled needle and syringe.

[2] Even the SoloShot FX™ autodisable syringe (**Chapter 7**) can be decontaminated because after use about 0.1 mL of chlorine solution can be drawn up. This volume is sufficient to completely fill the needle as well as cover the surface of the plunger and bottom of the syringe.

STEP 5: Remove the needle from the syringe. (If available, use forceps and grasp the needle at the base where it attaches to the syringe and carefully remove it by turning.)

STEP 6: Dispose of needle in a puncture-resistant sharps container. (When the container is three-quarters full, seal and either burn, encapsulate or bury it.)

STEP 7: Take the syringe apart, then wash in soapy water and rinse it at least three times with clean water.

STEP 8: Sterilize syringes by autoclaving or high-level disinfect them by boiling or steaming.

STEP 9: Store sterile or high-level disinfected syringes in a sterile or high-level disinfected container with a tight-fitting cover.

Reuse of Both Needle and Syringe (not recommended)

STEP 1: Do not recap the needle or disassemble the needle and syringe.

STEP 2: Immediately after use, fill the syringe with a 0.5% chlorine solution by drawing it into the syringe through the needle.

STEP 3: Decontaminate the assembled needle and syringe by placing in a 0.5% chlorine solution for 10 minutes.

STEP 4: Wearing utility gloves, remove from decontamination solution and push out (flush) solution from assembled needle and syringe.

STEP 5: Use forceps to take needle and syringe apart, then clean with soapy water. (Be sure to clean hub area of the needle.) Insert a stylet or needle wire through the hub of the needle to be sure it is not blocked.

STEP 6: Use forceps to put the syringe and needle back together. Rinse at least three times by filling with clean water and pushing out (flushing) water into another container so as not to contaminate the rinse water.

STEP 7: Use forceps to detach the needle from the syringe.

STEP 8: Examine the needle and syringe for:

- bent needle tip or other damage,
- needle hub fit to syringe, and
- readable syringe markers (lines indicating volume—cc or mL).

STEP 9: Use forceps to handle and dispose of damaged needles in a puncture-resistant sharps container. (When container is three-quarters full, seal and either burn, encapsulate or bury it.)

STEP 10: Sterilize by autoclaving or high-level disinfect needles and syringes by boiling or steaming.

STEP 11: Store disassembled sterile or high-level disinfected needles and syringes in a sterile or high-level disinfected container with a tight fitting cover.

REFERENCE

Kane A et al. 1999. Transmission of hepatitis B, hepatitits C and human immunodeficiency viruses through unsafe injections in the developing world: Model-based regional estimates. *Bull World Health Organ* 77(10): 801–807.

Phillips I et al. 1971. *Pseudomonas cepacia (multivorans)* septicaemia in an intensive care unit. *Lancet* 1(695): 375–377.

Simonsen L et al. 1999. Unsafe injections in the developing world and transmission of bloodborne pathogens: A review. *Bull World Health Organ* 77(10): 789–800.

APPENDIX F

CHEMICAL DISINFECTANTS

A number of chemicals are classified as high-level disinfectants and are used alone or in various combinations to disinfect medical instruments and equipment. In addition, some, such as glutaraldehydes and formaldehyde, are also classified for use as chemical sterilants ("cold sterilization") when items are soaked in them for prolonged periods of time (10–24 hours) (see **Chapter 11**).

Two new disinfectants, ortho-phthalaldehyde and peracetic acid, are not covered in this appendix. Experience with both is limited (e.g., peracetic acid is only used in the US in an automated machine for sterilization), and they are expensive. Rather, the intent is to provide performance characteristics on those disinfectants most readily available in countries with limited resources. These include chemicals such as alcohol (ethyl and isopropyl), chlorine and chlorine-releasing compounds, formaldehyde, various glutaraldehydes, and iodines and iodophors.

An exception to this is a novel new disinfectant, called superoxidized water, that has promise for use in developing countries. It is produced by electrolyzing saline (sea water) to create a disinfectant or antiseptic. Because the basic materials (sea water and electricity) are cheap, and the end product (water) is not damaging to the environment, superoxidized water could become an important new disinfectant some day. Because it loses activity with time, usually it is generated at the point of use. Recently, however, when tested under clean conditions, superoxidized water was found to be effective in disinfecting endoscopes within 5 minutes, even when 48 hours old (Selkon, Babb and Morris 1999). Unfortunately, at present the equipment needed to produce the product is expensive.

ALCOHOLS

Ethyl and isopropyl (2-propyl) alcohol (60–90%) are excellent disinfectants that are commonly available and inexpensive. Their rapid killing action and lack of chemical residue makes them ideal for disinfection of many medical items. The activity of both alcohols, however, drops sharply when diluted below 50%, with the optimal concentration range being 60–90% solutions with water (volume/volume).

In many countries, alcohol is available as "industrial methylated spirit" or ethyl alcohol denatured with a small amount of wood (methyl) alcohol (Harpin and Rutter 1982). Because methyl alcohol is the least effective alcohol, it should not be used alone as an antiseptic or disinfectant. Before using, be sure the ethyl alcohol is of adequate strength (60–90%) in locally available "spirit."

Ethyl and isopropyl alcohol are **not** considered to be high-level disinfectants because they do not inactivate bacterial endospores and some viruses. For example, isopropyl alcohol also does not kill hydrophilic viruses (e.g., echovirus, coxsackie virus) (Rutala 1996; Rutala 1993). Alcohols are, however, effective against HBV, HCV and HIV.

Advantages

Rapidly kill all fungi and bacteria including mycobacteria; isopropyl alcohol kills most viruses, including HBV and HIV, and ethyl alcohol kills all viruses; both are tuberculocidal (Rutala 1996).

- Rapid killing action.
- **Not corrosive** to metal.
- Inexpensive in comparison to other disinfectants.
- Useful for soaking rubber or latex items **occasionally**.
- Leave no chemical residue and therefore do not require rinsing.

Disadvantages

- Evaporate rapidly, which makes extended contact times difficult unless the items are immersed.
- Do not penetrate organic material and are easily inactivated.
- Flammable.
- May swell or harden rubber and plastics if used repeatedly or for prolonged periods of time.
- Damage shellac mounting of lenses in endoscopes.

Considerations for Use

- Primarily used as antiseptic and as low- or intermediate-level disinfectant (wiping oral and rectal thermometers and disinfecting external surfaces of equipment—stethoscopes, cryoprobe tips, ultrasound probes, Ambu bags or anatomic models).
- Store in a cool, well-ventilated area because they are flammable.

CHLORINE AND CHLORINE-RELEASING COMPOUNDS

Hypochlorites are the most widely used of the chlorine disinfectants and are available in liquid (sodium hypochlorite) and solid (calcium hypochlorite and sodium dichloroisocyanurate) forms.

Chlorine solutions and compounds are **high-level disinfectants** because they inactivate all bacteria, viruses, fungi, parasites and some spores (Russell, Hugo and Ayliffe 1982). They are fast-acting, very effective against HBV, HCV and HIV/AIDS, inexpensive and readily available. They are extremely useful for decontaminating soiled surgical instruments, gloves and other items as well as large surfaces such as examination tables (Shapshak et al 1993).

When potable (clean) water is available, 0.1% chlorine solution is satisfactory for high-level disinfection. If the chlorine is to be diluted with contaminated (unfiltered) tap water, a higher concentration (0.5%) should be used because much of the chlorine will be inactivated by the microscopic organic matter in the water (WHO 1988). (Instructions for preparing 0.1% and 0.5% chlorine solutions from liquid household bleach [sodium hypochlorite] are listed in **Table 10-1**.) Dilute solutions can also be made from other chlorine-releasing compounds available in powder (calcium hypochlorite or chlorinated lime) or tablet form (sodium dichloroisocyurate) (**Table 10-2**). If stored in closed brown bottles, various concentrations of commercial bleach solutions (1:100 to 1:5) do not lose their efficacy as fast as formerly thought (50% to 97% potency at 30 days), with higher concentrations being more stable (Rutala et al 1998).

Sodium Hypochlorite (Chlorine Bleach)

Advantages

- Usually is the least expensive and most readily available disinfectant.
- Easy to prepare and use.
- Quickly inactivates all viruses including HBV, HCV and HIV, as well as killing tubercle bacillus.
- Very useful for decontaminating soiled surgical instruments, gloves and other items, and large surface areas. (HLD takes 20 minutes, but decontamination can take as little as 60 seconds to kill HIV!)

Disadvantages

- Inactivated by organic matter. (Chloramine-T, an alternative compound that releases chlorine, is not inactivated by organic matter to the same extent as hypochlorites according to WHO 1988.)
- Loses potency on standing if left in **open** container (replace at least daily).
- May corrode metal instruments with prolonged exposure (>20 minutes) to concentrations greater than 0.5%. To minimize corrosion:
  - solutions should not be prepared or kept in metal containers (use plastic containers when possible);[1]
  - exposure time should not exceed 20 minutes; and
  - metal items should be thoroughly rinsed with water and dried after decontamination, or they can be placed in clean water for up to 1 hour before washing.

Note: Depending on the use, HLD versus decontamination, 0.1 % solutions can be made with boiled and filtered (if necessary) water in advance and stored in **closed** dark bottles for at least a month with little loss of potency.

[1] Electrolytic corrosion occurs when two or more dissimilar metals are placed in water or salt solutions, especially if the items are actually touching each other. To avoid this type of corrosion, steel and aluminum instruments should be immersed in separate trays. Also, if metal trays or pans (e.g., stainless steel) are used, a plastic mat or gauze pad should be placed on the bottom of the tray to prevent metal-to-metal contact during soaking. This is especially important when metal instruments are soaked for prolonged periods (12–24 hours) for chemical sterilization.

Considerations for Use

- Decontamination of surgical instruments, gloves and other items before cleaning (0.1%–0.5% depending on the quality of the water).
- HLD of plastic items, such as suction cannulae (0.1% made up in water that has been filtered and then boiled for 20 minutes). (See **Table F-1** for details.)
- Cleaning up blood or other potentially infectious body fluid spills and wiping down large surfaces (0.5%).
- Clean water for drinking or medical use (cleaning instruments) at 0.001%.

Calcium Hypochlorite or Chlorinated Lime

Calcium hypochlorite and chlorinated lime are available in powder form. Recommended dilutions are listed in **Table 10-2**.

- Calcium hypochlorite contains approximately 70% available chlorine.
- Chlorinated lime contains approximately 35% available chlorine.

The availability of prediluted chlorinated lime solutions can be confusing. For example, Eusol® is chlorinated lime and boric acid and contains 0.25% available chlorine. This is sufficient for disinfection of clean equipment, but is half the level recommended by WHO for decontamination of contaminated equipment (WHO 1988).

Advantages

- Both decompose more slowly than sodium hypochlorite, but they still should be protected by storing away from heat and light.

Disadvantages

- Inactivated by organic matter.
- Like all chlorine compounds, may corrode metal with prolonged exposure (>20 minutes) to concentrations greater than 0.5% unless thoroughly rinsed.
- More difficult to prepare dilute solutions due to poor solubility in alkaline water (pH >8) and amount of nondissolvable particulate matter in most products.

Sodium Dichloroisocyanurate

Sodium dichloroisocyanurate (NaDCC) forms hypochlorous acid when dissolved in water. It is available as powder or tablets. NaDCC powder has 60% available chlorine; NaDCC tablets contain 1.5 g available chlorine per tablet. (See **Table 10-2** for how to make recommended dilutions.)

Advantages

- NaDCC does not decompose as quickly as sodium or calcium hypochlorite.
- Tablets are easy to use for measuring.

Disadvantages

- More expensive than sodium or calcium hypchlorite.
- Like all chlorine compounds, they may corrode metal with prolonged exposure (>20 minutes) to concentrations greater than 0.5% unless thoroughly rinsed.

FORMALDEHYDE

Formaldehyde in both liquid and gaseous forms can be used as a chemical sterilant, as well as a high-level disinfectant (Taylor, Barbeito and Gremillion 1969; Tulis 1973). Its uses are limited by its irritating fumes and pungent odor. Formaldehyde is classified as a potential carcinogen; therefore, care must be taken to protect staff when preparing and using formaldehyde solutions (see **Disadvantages**, below).

A commercially available solution of formaldehyde (formalin), which contains 35–40% formaldehyde by weight, should be diluted with boiled water (1:5) to a final solution containing about 8% formaldehyde.

Details for preparing and using formaldehyde (formalin) solutions are provided in **Table F-1**.

Advantages

- Not readily inactivated by organic materials.
- Can be used for up to 14 days.
- Can safely be used on surgical endoscopes (laparoscopes) because 8% formaldehyde will not corrode metal or damage lensed instruments, plastics or rubber.

Disadvantages

- Causes skin irritation.
- Irritates the skin, eyes and respiratory tract, even at low concentrations.
- For sterilization, 24-hour soaking in 8% formaldehyde solution kills all microorganisms, including bacterial endospores.
- Produces a dangerous gas (bis-chloromethyl-ether) when mixed with chlorine.

Considerations for Use

- Because of the potential carcinogenicity in humans and noxious fumes, liquid or gaseous formaldehyde should not be used for HLD or sterilization if other high-level disinfectants are readily available. In many developing countries, however, formaldehyde continues to be used because both liquid and solid forms (paraformaldehyde) are extremely inexpensive, readily available and have been used in hospitals and clinics for many years. Switching over to less toxic compounds, such as glutaraldehydes or other newer high-level disinfectants, is strongly recommended but difficult to implement because of the high cost of alternatives.
- Replace solution sooner than 14 days if cloudy.
- Handle with care. Gloves should be worn to avoid skin contact, eyes should be protected from splashes and exposure time should be limited.
- Use only in a well-ventilated area. (OSHA exposure standard for formaldehyde limits the 8-hour time-weighted average exposure to a concentration of 0.75 ppm [OSHA 1991].)
- **Thoroughly** rinse equipment with sterile water or boiled and filtered (if necessary) water at least **three times** after soaking.

GLUTARALDEHYDES

Glutaraldehydes are widely used for chemical sterilization and HLD of medical instruments. Aqueous solutions are acidic (pH < 7) and only when made alkaline are they activated. There are many types of glutaraldehydes available worldwide. The most commonly used is an alkaline-stabilized 2% glutaraldehyde available commercially as Cidex® or Cidex 7®. These chemicals, which are derivatives of formaldehyde, also are irritating and the fumes very unpleasant; therefore, they should be used only in well-ventilated rooms.

Because the stability and activity of glutaraldehydes vary considerably depending on how they are prepared and stored, the manufacturers' directions must be followed closely. In general, for effective HLD, instruments and other items should be soaked for 20 minutes, while for sterilization, instruments should be soaked for 10 hours (see **Table F-1** for additional information).

Remember: Do not dilute unless specified in the manufacturer's instructions.

Until 1991, glutaraldehyde products were available in alkaline, neutral or acid forms. Since then, reports have documented that neutral or alkaline glutaraldehydes have superior killing power and anticorrosive properties when compared with acid glutaraldehyde (Rutala 1996). As a consequence, beginning in 1991 acidic glutaraldehyde products were gradually removed from the market. Recently, however, a new diluted product containing 0.95% glutaraldehyde with 1.64% phenol/phenate has been cleared by the USFDA for HLD. The antimicrobial efficacy of this product, however, needs to be

independently validated before it can be recommended. Also, like all glutaraldehydes, it is expensive.

Further details for preparing and using glutaraldehydes are provided in **Table F-1**.

Advantages

- Not readily inactivated by organic materials.
- Generally can be used for up to 14–28 days (see **Table F-1** for details).
- Can safely be used on surgical endoscopes (laparoscopes) because they will not corrode metal or damage lensed instruments (endoscopes), plastics or rubber.

Disadvantages

- Can cause skin irritation or dermatitis with chronic exposure.
- Vapors are irritating to mucous membranes (eye, nose and mouth) and respiratory tract.
- Work best at room temperature (20–25°C or 68–77°F).
- Expensive.

Considerations for Use

- At present, best disinfectant for HLD and cold sterilization of medical instruments that are heat-sensitive.
- Replace solution sooner than 14 days if cloudy.
- Wear gloves and protective eyewear in case of splashes and sprays.
- Use only in a well-ventilated area.
- **Thoroughly** rinse equipment with sterile water or boiled and filtered (if necessary) water at least **three times** after soaking.
- Soaking for longer than 20–30 minutes may be required to kill mycobacterium in cold climates.

Note: Some brands can be used for longer periods of time, up to 28 days. Check the manufacturer's instructions (Rutala 1996).

IODINES AND IODOPHOR SOLUTIONS

Iodine solutions (1–3% aqueous or tincture) and iodophors (iodine complexed with an organic material) have been used primarily as antiseptics for many years.

Note: Iodophors manufactured for use as antiseptics are **not** effective for disinfecting inorganic objects and surfaces. Antiseptics have significantly less iodine (Rutala 1996). Be sure to check the label.

Aqueous iodine solutions can be easily made up, and they as well as iodophors are readily available in most countries. Povidone-iodine (PVI) is a commonly available iodophor, usually sold as a 7.5–10% solution (1% iodine). (For instructions on preparing an iodophor solution, see **Table F-1**.)

Iodophors are **not** high-level disinfectants because conclusive evidence is lacking that they are effective against bacterial endospores and some fungi. Also, pseudomonas species, a group of gram-negative bacteria, have been

known to multiply in iodophors (Favero 1985; Rutala 1993). They are generally nontoxic and nonirritating to skin and mucous membranes.

Advantages

- Do not cause deterioration or softening of plastic items if items are kept dry between soakings.
- Diluted solutions of iodine and iodophors are nontoxic and nonirritating (unless the person is allergic to iodine).
- Can be used for disinfection of blood culture bottles and medical equipment such as thermometers.

Disadvantages

- Iodine is an **oxidizing agent** (causes rust) and should be used only for high-quality stainless steel equipment or plastic materials.
- Like alcohol and chlorine, iodine and iodophors are inactivated by organic materials; therefore, only previously cleaned instruments should be placed in iodine or iodophor solutions.
- **Thoroughly** rinse equipment with sterile water or boiled and filtered (if necessary) water at least **three times** after soaking.
- Allergic reactions can occur to staff handling iodine solutions and iodophors.

Note: Iodophors must be **properly diluted** to be effective. Correctly diluted iodophors have more active killing power than full-strength iodophors due to the decreased availability of "free" iodine in the full-strength products.

Considerations for Use

- Primarily used as antiseptic for skin and mucous membranes (aqueous preparations only)
- 3% aqueous solutions can be used for decontamination, but must be made fresh daily

Table F-1. Preparing and Using Chemical Disinfectants

CHEMICALS FOR STERILIZATION OR HIGH-LEVEL DISINFECTION

Disinfectant (common solution or brand)	Effective Concentration	How to Dilute	Skin Irritant	Eye Irritant	Respiratory Irritant	Corrosive	Leaves Residue	Time Needed for HLD	Time Needed for Sterilization	Activated Shelf Life[a]
Chlorine	0.1%	Dilution procedures vary[b]	Yes (with prolonged contact)	Yes	Yes	Yes[c]	Yes	20 minutes	Do not use	Change every 14 days, sooner if cloudy.
Formaldehyde (35–40%)	8%	1 part 35–40% solution to 4 parts boiled water	Yes	Yes	Yes	No	Yes	20 minutes	24 hours	Change every 14 days, sooner if cloudy.
Glutaraldehyde (Cidex®)	Varies (2–4%)	Add activator	Yes	Yes (vapors)	Yes	No	Yes	20 minutes at 25°C[d]	10 hours for Cidex	Change every 14–28 days; sooner if cloudy.
Hydrogen Peroxide (30%)	6%	1 part 30% solution to 4 parts boiled water	Yes	Yes	No	Yes	No	20 minutes	Do not use	Change daily; sooner if cloudy.
CHEMICALS FOR DISINFECTION (alcohols and iodophors are not high-level disinfectants)										
Alcohol (ethyl or isopropyl)	60–90%	Use full strength	Yes (can dry skin)	Yes	No	No	No	Do not use	Do not use	If container (bottle) kept closed, use until empty.
Iodophors (10% povidone-iodine) (PVI)	Approximately 2.5%	1 part 10% PVI to 3 parts water	No	Yes	No	Yes	Yes	Do not use	Do not use	If container (bottle) kept closed, use until empty.

[a] All chemical disinfectants are heat- and light-sensitive and should be stored away from direct sunlight and in a cool place (< 40°C).
[b] See **Tables 10-1 and 10-2** for instructions on preparing chlorine solutions.
[c] Corrosive with prolonged (> 20 minutes) contact at concentrations > 0.5% if not rinsed immediately with boiled water.
[d] Different commercial preparations of Cidex and other glutaraldehydes are effective at lower temperatures (20°C) and for longer activated shelf life. **Always** check manufacturers' instructions.

Adapted from: Rutala 1996.

REFERENCES

Favero MS. 1985. Sterilization, disinfection, and antisepsis in the hospital, in *Manual of Clinical Microbiology*, 4th ed. Lennette EH et al (eds). American Society for Microbiology: Washington, DC, pp 129–137.

Harpin V and N Rutter. 1982. Percutaneous alcohol absorption and skin necrosis in a preterm infant. *Arch Dis Child* 57(6): 478.

Occupational Safety and Health Administration (OSHA). 1991. OSHA amends formaldehyde standard. *Occupational Safety and Health News*: 1.

Russell AD, WB Hugo and GA Ayliffe. 1982. *Principles and Practice of Disinfection, Preservation and Sterilization*. Blackwell Scientific Publications: Oxford, England.

Rutala WA et al. 1998. Stability and bactericidal activity of chlorine solutions. *Infect Control Hosp Epidemiol* 19(5): 323–327.

Rutala WA. 1996. APIC guideline for selection and use of disinfectants. *Amer J Infect Control* 24(4): 313–342.

Rutala WA. 1993. Disinfection, sterilization and waste disposal, in *Prevention and Control of Nosocomial Infections*, 2nd ed. Wenzel RP (ed). Williams & Wilkins: Baltimore, MD, pp 460–495.

Selkon JB, JR Babb and R Morris. 1999. Evaluation of the antimicrobial activity of a new superoxidized water, Sterilox®, for the disinfection of endoscopes. *J Hosp Infect* 41(1): 59–70.

Shapshak P et al 1993. Inactivation of human immunodeficiency virus-1 at short time intervals using undiluted bleach. *J Acquir Immune Defic Syndr* 6(2): 218–219.

Taylor LA, MS Barbeito and GG Gremillion. 1969. Paraformaldehyde for surface sterilization and detoxification. *Applied Microbiol* 17(4): 614–618.

Tulis JJ. 1973. Formaldehyde gas as a sterilant, in *Industrial Sterilization: International Symposium*. Briggs Phillips G and WS Miller (eds). Duke University Press: Durham, NC, pp 209–238.

World Health Organization (WHO). 1988. *Guidelines on Sterilization and High-Level Disinfection Methods Effective Against Human Immunodeficiency Virus (HIV)*. WHO: Geneva, AIDS Series 2.

APPENDIX G

INSTRUCTIONS FOR OPERATING AND MAINTAINING HIGH-PRESSURE STEAM STERILIZERS (AUTOCLAVES)[1]

There are three types of high-pressure steam sterilizers:

- Gravity displacement
- Prevacuum
- Flash

Gravity Displacement Sterilizers

Small (table-top) to intermediate size sterilizers are frequently used in clinics and physicians' offices (**Figure G-1**). Larger in-wall mounted units are the most common type of high-pressure steam sterilizer used in hospitals.

Figure G-1. Simplified Diagram of a Gravity Displacement Steam Sterilizer

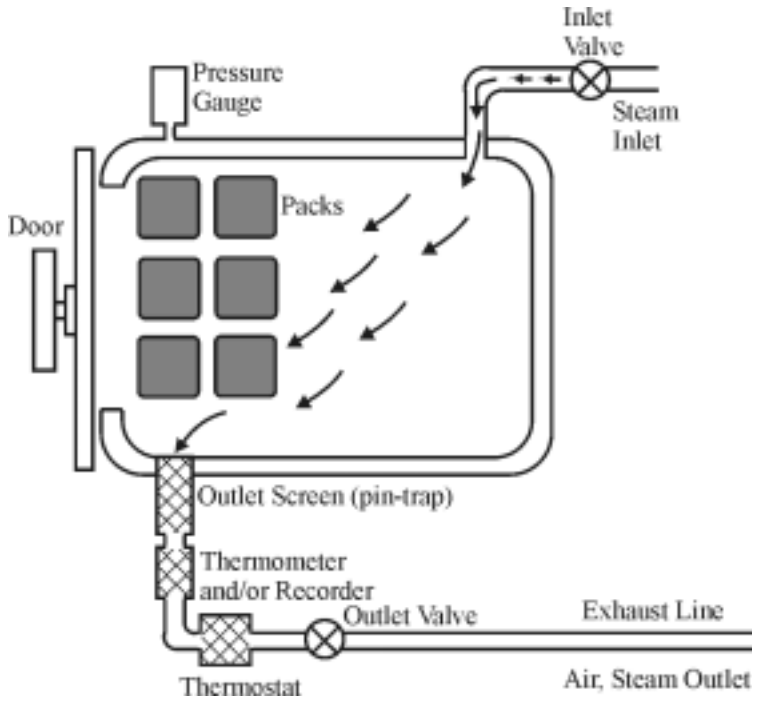


Table-top models are relatively simple to operate and essentially are horizontal pressure cookers. A pool of water in the bottom of the sterilizer is heated with electricity or kerosene until it turns into steam. The steam then rises to the top of the chamber because it is lighter than the cool air in the chamber. As more and more steam is produced, the cool air is forced out of the chamber through the drain near the bottom of the chamber. When the steam has pushed all the cool air out, steam will enter the drain, triggering the

[1] *Adapted from*: Tietjen, Cronin and McIntosh 1992.

thermally- (heat-) regulated valve to close. Once the valve is closed, the steam continues to build up pressure until the operating temperature (normally 121°C/250°F) is reached. The timer can now be activated and timing begun. At the end of the cycle (normally 20 minutes for unwrapped items and 30 minutes for wrapped items), the relief valve is opened which allows the steam to escape. Usually the steam passes through the water reservoir where it condenses back to water and thus does not enter the room. After the pressure on the gauge reads zero, the door can be opened 12–14 cm (5–6 inches). **Items should be left to cool for 30 minutes.** If steam is still present (and the chamber is quite warm), condensation of the moist air may cause wetness of the items or packs if they are placed on a cool or cold surface.

Remember: Small table-top (gravity displacement) sterilizers should not be confused with so-called office "sterilizers." These inexpensive "sterilizers" have a tray on which instruments are placed and when the lid is lowered the items are immersed in boiling water. They really are just boilers and can be used for HLD only.

This type of sterilizer should be checked routinely by running a biological indicator test (see **Chapter 12**). Also, whenever possible, it is recommended that temperature-specific indicators (as well as autoclave tape) be used with each cycle (Webb 1986).

Prevacuum Sterilizers

These sterilizers are similar to the gravity displacement sterilizers except that they have a vacuum pump system to remove the air in the chamber before the steam is let in. This step reduces the total cycle time. Most prevacuum sterilizers are operated at the same temperature (121°C/250°F) as gravity displacement sterilizers. A special type of vacuum sterilizer, called a high-speed vacuum sterilizer, however, is operated at a higher temperature, 134°C/275°F. The vacuum system not only shortens the cycle time, but also reduces the chance of air pockets from forming. Because a prevacuum sterilizer is more complex to operate, it is important to monitor its use closely and for it to be regularly maintained.

Flash Sterilizers

These are small, table-top prevacuum sterilizers, usually located in operating rooms or adjacent to them. They operate at a higher temperature (134°C/275°F) and thus have a shorter cycle time. Normally, because of their small size, their use is limited to sterilization of unwrapped surgical instruments for emergency purposes (e.g., dropped instruments, etc.).

In summary, in most healthcare facilities **gravity displacement sterilizers** are the type most frequently encountered. **High-speed vacuum** and **flash sterilizers** usually are found only in large referral hospitals in most countries (Webb 1986).

OPERATION

Instructions for operation and routine maintenance of steam sterilizers (autoclaves) should be included in the basic training of healthcare staff. A steam sterilizer will reliably sterilize items only when kept in good working condition and operated correctly.

Sterilization by steam requires four conditions: **adequate contact, sufficient temperature, proper time and sufficient moisture**. While all are necessary for sterilization to take place, sterilization failures in clinics and hospitals are most often caused by lack of steam contact or failure to attain adequate temperature (Webb 1986).

Contact

The most frequent reason for sterilization failure is the **lack of contact** between the steam and the microorganisms. This failure may be related to human error or mechanical malfunction. Frequent causes of steam contact failure include the following:

- **Failure to clean the object being sterilized adequately**. Any coating of soil can protect the microorganisms from direct steam contact. In addition, the effectiveness of sterilization is dependent on the "bioload" (number of microorganisms) present prior to the sterilization cycle.
- **Instruments closed, locked or stacked**. All instruments must be packed in an open and unlocked position, or disassembled, so that steam can reach all surfaces (e.g., Place gauze or linen in between bowls so that steam can reach all surfaces of each bowl).
- **Packages wrapped too tightly**. Air and steam do not mix readily. Air, being heavier than steam, normally is displaced to the bottom of the sterilizer and is then forced out through the drain. If the packs are wrapped too tightly, however, air is trapped and cannot escape. It forms cool air pockets at the center of the packages, preventing the items from reaching temperatures sufficient to kill all microorganisms.
- **Packs too crowded**. It is essential that the packs be arranged loosely on the cart or the same type of problem as that in the above example will occur. Packs should be placed on the edge because it is easier for air to be displaced downward between the packs than to go through the many layers of fabric of horizontally placed packs.
- **Wrong position of container**. If pans, bottles or other airtight containers are to be sterilized, including containers with instruments inside, it is essential that the tops be removed (or held loosely in place) and that the containers be placed on their sides. (If the containers are placed upright, the air cannot be displaced and will be trapped in them.)
- **Clogged strainer**. At the bottom of most sterilizers is a small drain strainer used to keep lint, pins and other small objects from entering the exhaust line. It is essential that these screens be cleaned daily, or they will become clogged and trap air in the sterilizer.
- **Other mechanical problems**. Several other problems can occur, such as a defective steam trap and clogged exhaust lines. Often, the sterilizer operator cannot repair such problems. In some cases, however, a weekly flush of hot liquid soap through the exhaust line will keep it cleaned out. If the sterilizer manual calls for this weekly flush, it must be performed or sterilization failure may occur.

From a review of the above, it is clear that most failures in sterilization begin with human error. By becoming familiar with these problem areas, staff responsible for operating the sterilizer can avoid the major causes of sterilization failure. To detect steam contact failures, the use of an internal (inside the package) indicator is strongly recommended.

Temperature

The next most important factor in steam sterilization is **temperature**. The most commonly used temperature for steam sterilization is 121°C (250°F). When an object at room temperature is placed in a sterilizer, the steam transmits thermal energy to the object until the object reaches the same temperature as the steam. Under normal conditions this equilibrium occurs within a few minutes. If the steam is unsaturated (too dry) or if the steam is prevented from reaching all parts of the object, the temperature may never reach the level required for sterilization. The **only** way to be certain the sterilizer is working correctly is to ensure that the temperatures **at all points inside the load** reach the full operating temperature of 121°C (250°F).

Note: The temperature must never be allowed to drop below 121°C/250°F. If this should happen, sterilization may not take place. (If available, a temperature specific indicator tape that changes color should be used to be sure that all items have been sterilized. When removing the pack, if the tape has not changed color, repeat the sterilization cycle.)

The temperature gauges and recorders located on the sterilizer control panel sense the temperature of the exhaust line and do not give an indication of center-of-pack temperature. While these sensing devices do give a good indication of overall sterilizer operation, they cannot detect air pockets within packs and similar problems.

Timing

Just as it takes a certain amount of time to cook food, **it takes a certain amount of time to kill microorganisms**. In both cases, the hotter the temperature, the less time is required. Sterilization time is measured in **D-values**. A **D-value** is the amount of time required to kill 90% of the microorganisms present. Different microorganisms are killed in different amounts of time so each kind of microorganism has a different set of D-values, and of course, the D-value depends on the temperature.

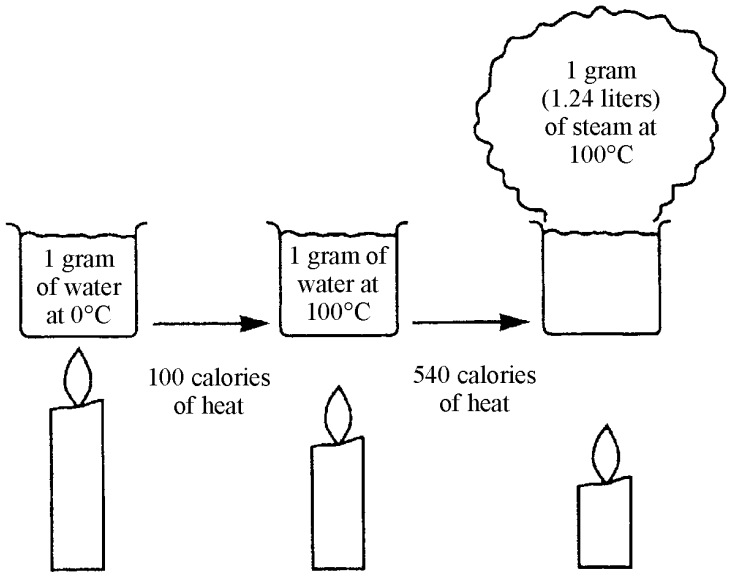
A highly resistant but relatively harmless (nonpathogenic) microorganism called *Bacillus stearothermophilus* is used to test steam sterilizers. As used in hospitals and clinics to test sterilizers, this microorganism has a D-value of about 2 minutes at 121°C (250°F). In other words, it would take 2 minutes at 121°C (250°F) to kill 90% of the test microorganisms present. Through research, mathematical calculation and intelligent "guesses," authorities have generally agreed that for normal hospital sterilization about six *Bacillus stearothermophilus* D-values (or about 12 minutes) should be sufficient to kill essentially all pathogenetic microorganisms and give a large margin of safety. Because in many countries internal temperature sensing devices, such as temperature-specific chemical indicators, are not available, **extra time is recommended as an added safety margin**. Twenty minutes for unwrapped and 30 minutes for wrapped packs are sufficient to kill most microorganisms.

Moisture

Last, but not least, is the **moisture** requirement. Adequate moisture content of the sterilizer atmosphere is mandatory for effective sterilization by steam. Adequate moisture content means that the steam must be "saturated," having a relative humidity of 100%. When any cool object is placed in the sterilizer, the steam at the surface of the object is cooled and becomes supersaturated. Water begins condensing on the surface of the object. This condensation produces two immediate effects:

- The volume of gas in the sterilizer chamber decreases as the steam (water vapor) changes to the liquid state and more steam is drawn into the chamber and into contact with the articles being sterilized.
- Very large amounts of thermal energy are transferred to the object, raising the temperature of the article significantly. The amount of heat released is best explained by comparing the calories required to change the temperature of steam as compared to the calories absorbed when water is converted to water vapor (steam) (**Figure G-2**).

Figure G-2. Calories of Heat, Water Temperature and Conversion to Steam



Source: Webb 1986.

One calorie of heat will raise the temperature of 1 gram of water 1°C. Thus, 100 calories are required to raise the temperature of 1 gram of water from 0°C to 100°C. To convert that same gram of water into steam (i.e., vaporize it), an additional 540 calories are required. When the steam condenses during the sterilization process, heat is transferred to the items being sterilized and the steam turns to water at 100°C.

If the steam is not saturated (less than 100% relative humidity), two problems will develop, either or both of which will interfere with the adequacy of the sterilization process:

- Articles in the sterilizer will remain dry, and any microorganisms present cannot be killed as readily as under wet conditions. (Water vapor softens the capsules of microorganisms, making them more vulnerable to destruction by heat.)
- Articles in the sterilizer will remain "cool" much longer, especially if they are wrapped. Again, using the home kitchen as an example, if a kettle of beans is placed in an oven (dry heat), it may take hours for them to be cooked. On the other hand, if they are placed in a pressure cooker (saturated steam), they will cook much more quickly. Saturated steam is a much better "carrier" of thermal energy than dry air.

In summary, saturation of the steam is vital to sterilizer operation because water vapor is the best carrier of thermal energy and because the vapor makes the microorganisms more vulnerable to destruction by heat (Webb 1986).

OPERATING INSTRUCTIONS (Gravity Displacement Steam Sterilizers)

To ensure correct operation, when available, **consult specific operating instructions supplied by the manufacturer.** (See **Figure G-1** for a simplified diagram of a gravity displacement steam sterilizer.) The following are general instructions that should be effective for most steam sterilizers:

STEP 1: Arrange packs in the chamber to allow free circulation and penetration of steam to all surfaces. In large sterilizers, which have carts, packs should be loaded onto the cart and then rolled into the sterilizer.

STEP 2: Sterilize wrapped objects for 30 minutes, unwrapped objects for 20 minutes. Use a timer to keep track of time. The temperature should be 121°C (250°F); the pressure should be 106 kPa (15 lbs/in^2).

STEP 3: Wait about 30 minutes (or until the pressure gauge reads zero) to permit the sterilizer to cool sufficiently before opening the lid to allow steam to escape. Open the door of the sterilizer a small amount and allow instrument packs to dry completely before removal; this may take an additional 30 minutes. (Damp packs act like a wick drawing in microorganisms from the environment.) Wrapped instrument packs are considered unacceptable if there are water droplets or visible moisture on the outside of the package when removed from the chamber.

STEP 4: To prevent contamination by condensation, place sterile trays and packs on a surface padded with paper or fabric after removing the packs from the chamber. (Do not store trays or packs until they reach room temperature; this usually takes about 1 hour.)

STEP 5: After sterilizing, objects wrapped in cloth or paper are considered sterile if kept dry and the package intact. Sealing packs in plastic bags can help to prevent damage to the packs and permits a longer shelf life. Unwrapped objects must be used immediately or placed in a sterile, covered container.

Problem solving

If steam escapes from the safety valve or under the lid, the autoclave is not working correctly and it is merely steaming items at low-pressure (HLD, not sterilization). What to do?

- If steam escapes from the safety valve instead of the pressure valve, **the pressure valve must be cleaned and inspected**.
- If steam escapes from under the lid, **the gasket (rubber ring) must be cleaned and dried or replaced**.

STEAM STERILIZATION PROCEDURE

Sterilization depends on correctly following certain practices and processes. These include:

- routine maintenance,
- preparing items to be sterilized,
- packaging and wrapping,
- loading,
- operating, and
- unloading the sterilizer.

Note: Only when all these procedures are done correctly will items be sterile.

Routine Maintenance

Although there are many brands of steam sterilizers, routine maintenance practices generally are the same regardless of the make or type. (See **Figure G-1** for a simplified diagram of a gravity displacement sterilizer.) For routine maintenance:

- The **outlet screen** (or **pin-trap**) should be removed daily and cleaned using a mild soap and brush under running water.
- The chamber should be cleaned daily using a soft cloth, or for large sterilizers, a long-handled mop which is used only for this purpose. Do **not** use abrasives or steel wool because they may scratch the stainless steel surface and increase the occurrence of corrosion.
- All door gaskets should be cleaned daily with a lint-free cloth and checked for defects. Defective rubber gaskets should be replaced.
- The carriage (loading cart used to hold the packs placed in a sterilizer) should be cleaned daily using a mild soap and lint-free cloth. (The wheels

Note: The chamber should be cooled before doing any procedure (e.g., loading or cleaning).

of the loading cart also should be cleaned at this time, removing any string or other debris.)

- The **exhaust line** (or chamber drain) should be flushed weekly. This will keep the drain free of substances that might hinder air or steam removal from the chamber. Before flushing the **exhaust line**, check the maintenance instructions because trisodium phosphate solution (a special type of soap) often is recommended (DHEW 1975; Webb 1986). This can be prepared by adding 1 ounce trisodium phosphate to 1 liter (1 quart) hot water. If this chemical is not available, the **exhaust line** can be flushed with hot water containing a mild soap solution. To do this, first remove the screen. Then pour 1 liter (1 quart) of the solution down the drain using a funnel. Complete the process by pouring a liter of hot water to rinse out the soap and replace the screen.

Because the specific operating instructions for a high-pressure steam sterilizer (autoclave) usually also contain instructions for routine maintenance, managers should make copies of these instructions available for staff to use. If replacement copies are needed, they can be obtained by writing to the individual manufacturer (normally the address can be found on the autoclave) or from the donor agency providing the equipment.

Preparing Items for Steam Sterilization

All instruments and other items should be decontaminated and thoroughly cleaned and dried before being sterilized. In some cases, it is not necessary to completely dry the items being sterilized, such as needles or other items with small openings because the small amount of water left inside helps in the steam sterilization process. For such items, after cleaning, flush with distilled or boiled water just prior to packaging for steam sterilization. Finally, all jointed instruments should be open (or in the unlocked position) and disassembled. Reusable cloth items should be laundered and dried after use or prior to sterilization in order to:

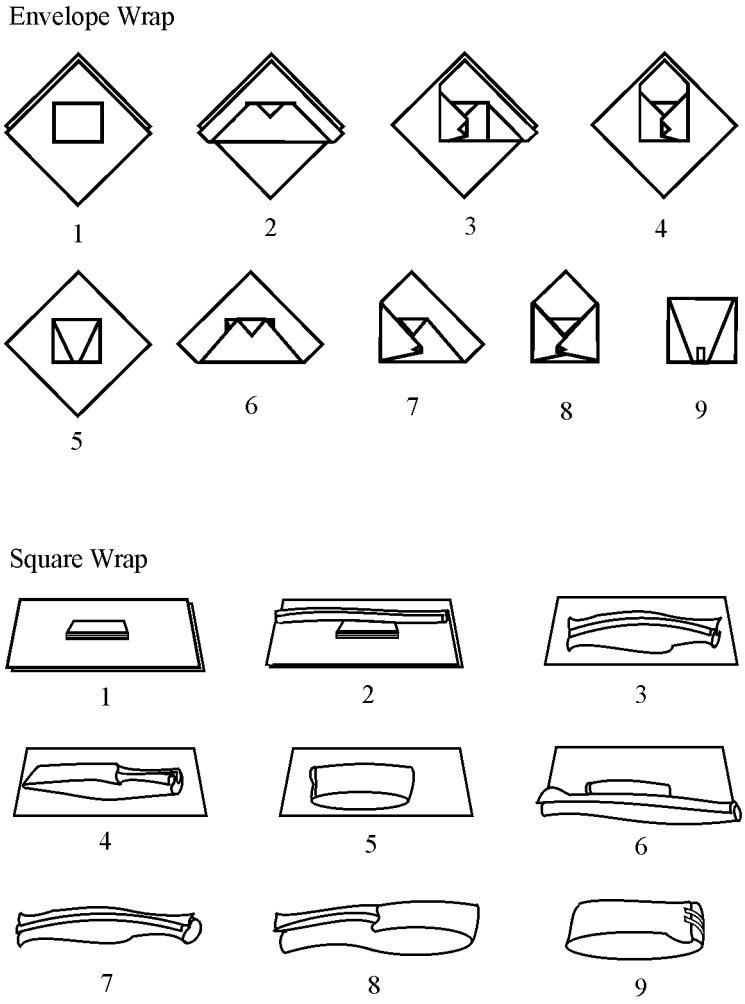
- remove organic matter, and
- prolong the life of the cloth by restoring the fabric's normal moisture (water) content.

Packing and Wrapping

Wrapping items to be sterilized permits sterile items to be handled and stored without being contaminated. (See **Figure G-3** for examples of typical wrapping techniques.) Materials used for wrappers should:

- Allow air removal and steam penetration
- Act as a barrier to microorganisms and fluids
- Resist tears and punctures and be free of holes
- Be nontoxic and low-lint
- Be inexpensive

Figure G-3. Typical Wrapping Techniques



Types of materials that can be used as wrappers include:

- **Muslin cloth** (140 thread count): Use **two** double thickness wraps (four layers in all), as this is the least effective of the materials used for wrapping. Use for both steam and dry heat sterilization.
- **Paper**: Double wrapping (two layers) recommended. Use for steam sterilization only and **do not reuse**.

Do not wrap items in any waterproof material, such as plastic or canvas, for steam sterilization, as steam will not penetrate the material and the item will not be sterile.

Wrappers should not be reused if they are torn, stained with oils or if they have hard or gummy deposits. Linen wrappers should be laundered between sterilizations, **even if unused**, in order to restore moisture to them (dried out fibers decrease the ability of the cloth to form a barrier to microorganisms). Dust covers (sealed plastic bags 2–3 mils thick) can protect the integrity of

sterile packs during storage. Packs should be placed in plastic bags or other dust covers after cooling.

Tips for Wrapping

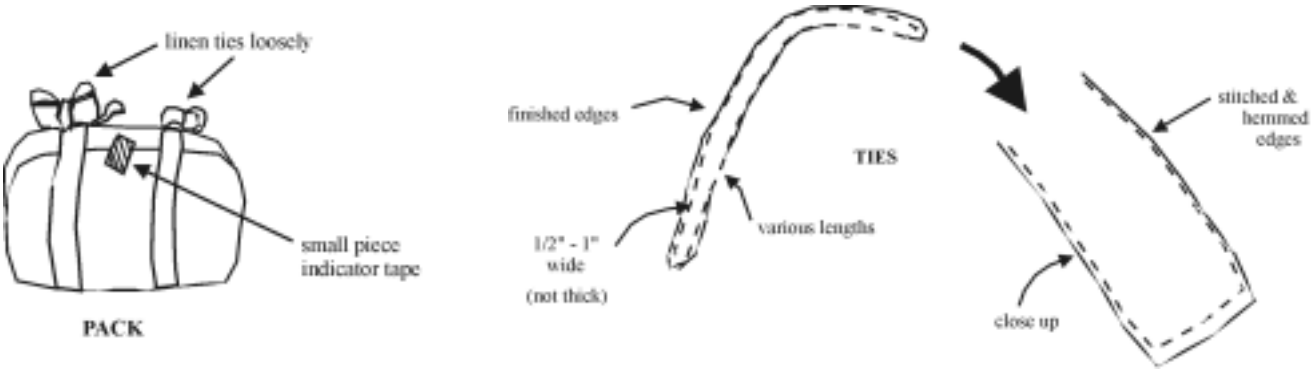
At least two layers of wrapping should always be used to reduce the possibility of contaminating the contents during unwrapping.

Do **not** wrap packages too tightly. If wrapped too tightly, air can become trapped at the center of packages, preventing the temperature from getting high enough to kill all the microorganisms. Also, wrapping with strings or rubber bands or **tying linen ties too tightly** can prevent steam from reaching all surfaces.

The outer wrapper of the pack can be loosely secured using linen **ties** (as described below) or masking tape. (The use of indicator tape for holding packs together should be minimized as it is expensive and very hard to remove from linen. It is best used in the center of the pack to verify steam penetration.)

Packs can be secured with linen **ties** made from the same cloth. Hemmed strips about ½ inch wide, in various lengths, can be used one or two to a package and eliminate the need for a lot of expensive and hard-to-remove indicator tape. They can be used to secure almost any size package (see **Figure G-4**).

Figure G-4. Packs and Ties



Loading and Unloading

O*bjectives*

- To load items into the autoclave in such a manner as to allow passage of the most steam through the load.
- To unload the steam autoclave in such a manner as to maintain the sterility of the items that have been processed through a sterilizing cycle.

General Principles

- When loading, leave sufficient space for steam to circulate freely. **Do not overload.**
- Place all packs (linen, gloves) on edge and place canisters, utensils and treatment trays on their sides.
- Place instrument sets in trays having mesh or perforated bottoms flat on the shelves.
- In combination loads of cloth (or paper) packs and instruments trays, place linens on top shelves and trays on lower shelves. This prevents any condensation (moisture), which forms on cool metal when steam initially contacts the item, from dripping onto linen packs (DHEW 1975).
- Surgical gloves should be sterilized by themselves or placed on the top shelves.
- Nested packs should be positioned in the same direction to help prevent air pockets, so condensation can drain and steam can circulate freely.
- Shelves (metal wire) or a loading cart must be used to ensure proper loading. It is preferable to use the cart that comes with the sterilizer.

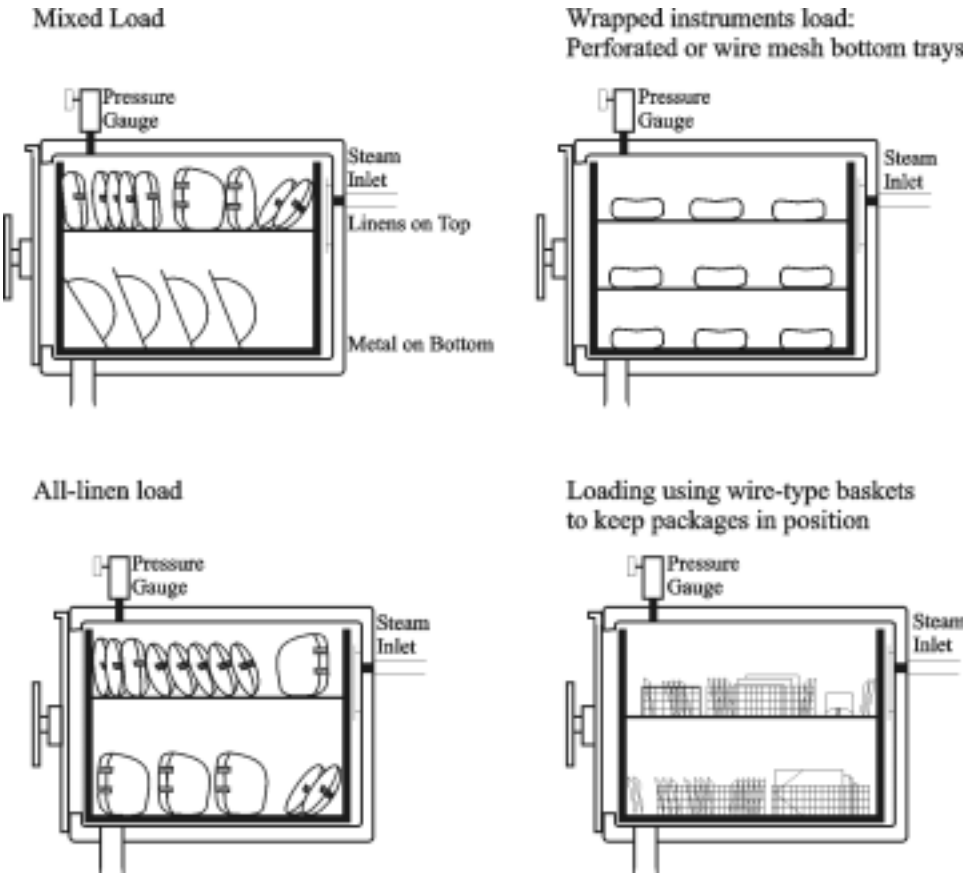
See **Figure G-5** and **Tables G-1** and **G-2**.

Remember: **If an item goes in wet, it will come out wet.** All items (instruments, basins and glassware) must be dry before loading into the sterilizer. This helps prevent "wet packs." The sterilizer is capable of drying items that have become moist during a properly loaded and operated sterilization process, but it **cannot** remove excess moisture.

Metals and Glassware

- Instrument sets should not exceed 8 kg (18 lbs). Basin sets should not exceed 3 kg (7 lbs). This is to limit the amount of condensation which forms when steam contacts cool metal. Using these limits ensures that the items will dry during the sterilization cycle.
- Solid containers should be **placed on their sides** to allow airflow out of them. If air is trapped in a solid container, it will prevent the steam from contacting the inner surface and prevent sterilization.

Figure G-5. Loading a Steam Sterilizer



Source: AAMI 1990.

Surgical Gloves

Sterilize in separate loads (see **Appendix C** for step-by-step instructions).

Linens

- Linen packs should not be too large and weigh no more that 5 kg (12 pounds) in order to assure steam penetration of the pack in 30 minutes (the time allowed for sterilizing wrapped items).
- Packs containing sheets, table covers and towels are the most difficult for steam to penetrate and contact each fiber. Such packs must be placed on edge on the shelf to insure steam penetration.

Liquids

- Liquids must be sterilized by themselves.
- The amount of liquid in the bottle, not the size of the container, determines the time required for sterilization.

- Use only Borosilicate heat-resistant glass (Pyrex®).
- Use only automatic self-sealing caps for closure.

Table G-1. Loading the Steam Sterilizer Using Loading Carts or Shelves

ESSENTIAL STEPS	KEY POINTS
Place all items on a shelf. Use either a loading cart or shelves in the sterilizer.	Never place items (wrapped or unwrapped) on the floor of the sterilizer. Items placed on the floor could block discharge of air from the sterilizer, or allow air and moisture to be trapped in pockets, resulting in sterilization failure and "wet packs."
Items must not touch chamber walls.	Packs touching the chamber walls can be scorched or contents damaged due to excessive heat of the metal walls.
Always allow 7–8 cm (3 inches) of space between top-most package and top of chamber.	This allows displacement of air and free flow of steam.
Place all fabric packs on the edge (folds perpendicular to shelf); and when loading two layers on one shelf, place the upper layer crosswise to the bottom layer.	It is easier for steam to flow down through the folds to penetrate each fiber than through flat, compressed surfaces.
Place all bottles, solid metal and glass containers of dry materials on their sides with lids held loosely in place.	Air will drain out and steam will take its place.
Place treatment trays and utensils on the edge, tipped slightly forward.	This prevents pooling of condensation and facilitates drying.
Place instrument trays (mesh or perforated bottom only) flat on shelves. If instruments have been placed in a solid tray or on a Mayo tray, the tray must be placed on the edge and tipped slightly forward.	This helps maintain an orderly arrangement of contents and reduces damage caused by "dumping" all the instruments into bottom of tray if instrument tray is placed on its side. This also facilitates drying.
Solutions must be sterilized by themselves, and placed on the shelves not touching each other.	There is always a possibility that solutions will explode. If instruments and other items are in the steam sterilizer, they will be contaminated and they may be damaged.
Use a wire basket to hold glove packages upright. Never place packages on top of each other.	If gloves are stacked, the compression at the bottom of the pile will prevent access of steam to the gloves.
Use only the upper shelves for gloves. Place glove packages loosely on edge with thumbs up, well away from the walls of the chamber. **Never** place them on the bottom shelf of the chamber.	Residual air gravitates to the lower part of the chamber and will increase the rate of deterioration of the rubber.
Do not compress packages or overload the chamber.	When placing packages on shelves, put hand between them to be sure packages are not compressed and give least possible resistance to steam throughout the load.

Adapted from: Tomlinson et al 1996.

Combination Loads

- In loads which combine linens (fabrics) and metal items, place linens on top shelves and metal items below. This prevents condensation from dripping onto the linen packs, causing them to absorb the excess moisture.
- When a load is made up of wrapped and unwrapped items requiring different times to ensure sterilization, **the longest required time (i.e., 30 minutes) must be used**.

Remember: The sterilizer is unable to remove excess moisture.

The fundamental rule in loading the sterilizer is to prepare all items and to arrange the load in such a manner as to present the least possible resistance to the passage of steam through the load (i.e., from the top of the chamber toward the bottom).

Unloading Tips

- Allow instrument packs to dry completely before removal (takes 30 minutes).
- Place sterile trays and packs on surfaces padded with paper or fabric. (Do not place warm packs on cold metal surfaces, as condensation will occur.)
- Store when packs reach room temperature (usually takes about an hour).
- Sterilized packs and articles should be handled gently and as little as possible.

Note: If a pack is dropped, tears or comes in contact with moisture, it must be considered contaminated.

Table G-2. Unloading the Steam Sterilizer

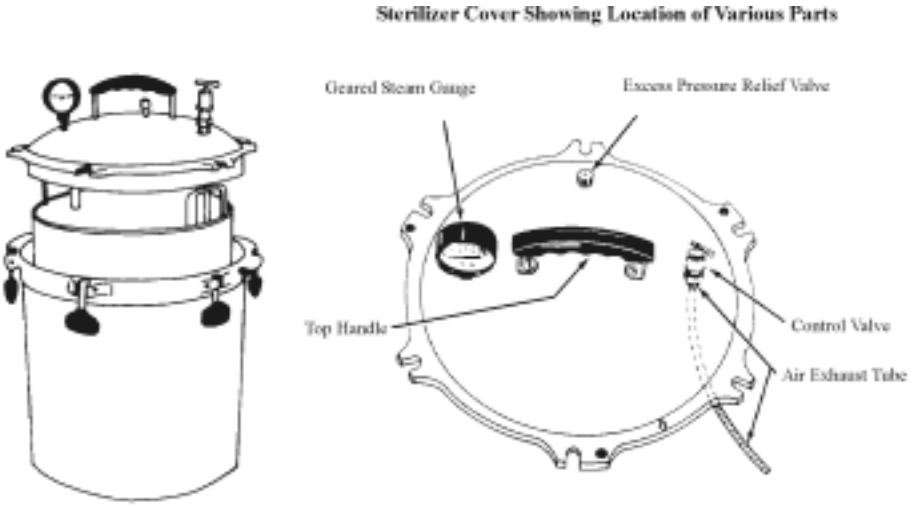
ESSENTIAL STEPS	KEY POINTS
After the sterilizing cycle has been completed and the chamber pressure gauge reaches "0," open the door 12–14 cm (5–6 inches).	Always keep the door between you and the sterilizer when opening the door.
Wait 30 minutes before unloading the sterilizer.	This allows residual moisture to dry and the load to cool.
Unloading Using a Loading Cart	
Remove the loading cart from the sterilizer and place it where there is no open window or fan in close proximity.	Do not place freshly sterilized packages, especially those not completely cooled, in front of an open window or a fan because there may be residual humidity in the packages, and dust and dirt could be forced through the wrappers, contaminating the contents. If there are water droplets or visible moisture on the outside of the wrapper or package, or on the tape used to secure it, the package is contaminated. It must be reprocessed before use.
Look at, but do not handle, the outside of the package to test for dryness.	
When the packs reach room temperature, remove packs from the loading cart and place on storage shelves. They may be dispensed or placed in sterile storage.	It may take 1 hour or longer for packs to reach room temperature. Avoid unnecessary handling.
Unloading Using Shelves (loading cart not used)	
Remove packages from the sterilizer shelves.	Avoid unnecessary handling.
Look at outside of the wrappers for dryness..	If there are water droplets or visible moisture (water stains) on the outside surfaces of packages, or on the tape used to secure it, the package is contaminated. It must be reprocessed before use.
Place packs on a surface well padded with paper or fabric, away from open windows or the front of a fan.	In order to prevent condensation from forming, do not place on a cool or cold surface. Do not place freshly sterilized packages, especially those not completely cooled, in front of an open window or a fan because there may be residual humidity in the packages, and dust and dirt could be forced through the wrappers, contaminating the contents. If there are water droplets or visible moisture on the outside of the wrapper or package, or on the tape used to secure it, the package is contaminated. It must be reprocessed before use.
When packages have cooled to room temperature, they may be dispensed or placed in sterile storage.	It may take 1 hour or longer for packs to reach room temperature. Avoid unnecessary handling.

Adapted from: Tomlinson et al 1996.

GENERAL INSTRUCTIONS FOR OPERATING GRAVITY DISPLACEMENT NONELECTRIC (PRESSURE COOKER TYPE) STEAM STERILIZER

The steam sterilizer should be run at 121°C (250°F) 106 kPa (15 lbs/in^2) for **20 minutes** for unwrapped items, **30 minutes** for wrapped items. As moist heat is the sterilizing agent (i.e., it kills the microorganisms), the temperature gauge on the exhaust line should be used to monitor when to begin timing the sterilization cycle, not just the pressure gauge alone (DHEW 1975).

Figure G-6. Gravity Displacement Sterilizer (Nonelectric)



To ensure correct operation, consult specific operating instructions supplied by the manufacturer, when available.

Instructions

STEP 1: Decontaminate, clean and dry all instruments to be sterilized.

STEP 2: Put all jointed instruments in the opened or unlocked position. Disassemble instruments composed of more than one part or sliding parts. To help prevent dulling of sharp points and cutting edges, wrap the sharp edges and needle points in gauze.

STEP 3: Wrap clean instruments or other objects in a double thickness of muslin or paper wrap. Instruments should not be held tightly together by rubber bands or any other means that will prevent steam contact with all surfaces.

STEP 4: Arrange packs in the chamber to allow free circulation and penetration of steam to all surfaces.

STEP 5: When using a pressure cooker or kerosene-powered sterilizer, bring water to boil until steam escapes from the **pressure valve** only; turn down heat but keep steam coming out of pressure valve. Do not allow to boil dry. Steam should always be escaping from the pressure valve.

STEP 6: Sterilize for 30 minutes for wrapped objects, 20 minutes for unwrapped objects. Time with a clock. The temperature should be 121°C (250°F); the pressure should be 106 kPa (15 lbs/in^2).

STEP 7: Wait about 30 minutes (or until the pressure gauge reads zero) to permit sterilizer to cool sufficiently before opening the lid to allow steam to escape. Allow instrument packs to dry completely before removal; this may take an additional 30 minutes. (Damp packs act like a wick drawing in microorganisms from the environment.) Wrapped instrument packs are considered unacceptable if there are water droplets or visible moisture on the outside of the package when removed from the sterilizer chamber.

STEP 8: To prevent contamination by condensation, place sterile trays and packs on a surface padded with paper or fabric after removing from the chamber. (Do not store trays or packs or place in a plastic dust cover until they reach room temperature; this usually takes about 1 hour.)

STEP 9: After sterilizing, objects wrapped in cloth or paper are considered sterile if kept dry and the package intact. Sealing packs in plastic bags can help to prevent damage to the packs and permits a longer shelf life. Unwrapped objects must be used immediately or placed in a sterile, covered container.

STEAM STERILIZING LIQUIDS

Sterile water can **only** be prepared by steam sterilization.

Instructions

- All liquids should be in heat-resistant glass (Pyrex), closed with automatic self-sealing caps.
- Load steam sterilizer with liquids only.
- Wait until thermometer indicator shows 121°C (250°F) and 106 kPa (15lbs/in^2).
- Time the sterilization using a clock. The amount (volume) of solution in the bottle determines the sterilization time, **not the size of the bottle**:

75–200 ml	20 minutes
200–500 ml	25 minutes
500–1000 ml	30 minutes
1000–1500 ml	35 minutes
1500–2000 ml	40 minutes

Note: If bottles of solutions with different volumes are sterilized in the same load, use the sterilization time recommended for the bottle containing the largest volume of liquid.

- When the sterilization cycle has ended, release the pressure slowly, taking **not less than 15 minutes,** until the chamber pressure is at "0." Turn operating valve off and open the door **only 1 cm (½ inch)**. (Suddenly opening the door all the way after a sterilization cycle could cause liquids to boil over or bottles to burst.) Wait an additional 30 minutes for the chamber to cool before removing the load.

REFERENCES

Association for the Advancement of Medical Instrumentation (AAMI). 1990. *Standards and Recommended Practices Volume 2: Sterilization*. Arlington, VA, pp 148–149.

Department of Health, Education and Welfare (DHEW). 1975. *A Manual for Hospital Central Services*. DHEW # HRA 75-4012, pp 80–83.

Tietjen LG, W Cronin and N McIntosh. 1992. Autoclaves, in *Infection Prevention Guidelines for Family Planning Programs*. Essential Medical Information Systems, Inc.: Durant, OK, pp 208–229.

Tomlinson MJ et al. 1996. Infection Control Techniques/Procedure Manual. Chosen Mission Project: Erie, PA.

Webb SB (ed). 1986. *Central Service Technical Manual*, 3rd ed. International Association of Hospital Central Service Management (IAHCSM): Chicago.

APPENDIX H

DECONTAMINATION, CLEANING, HLD AND CHEMICAL STERILIZATION OF LAPAROSCOPES

Surgical endoscopes (laparoscopes) are delicate instruments that must be handled with great care to prevent damage. The following recommendations will help to protect these instruments and prolong their use. Laparoscopes and accessories should be sterilized or high-level disinfected using chemical agents. Glutaraldehyde and formaldehyde are the best chemical high-level disinfectants for soaking laparoscopic instruments because they do not damage rubber, plastics or lens cements. Other high-level disinfectants, such as 6% hydrogen peroxide, may be corrosive.

HOW TO DECONTAMINATE AND CLEAN LAPAROSCOPES AFTER USE[1]

Note: Because alcohol rapidly kills HBV and HIV, this step protects handlers against possible hepatitis B and AIDS infection.

STEP 1: Immediately after use, gently wipe the laparoscope, fiber-optic light source and cable and plastic tubing with Luer-Lok™ with a cloth soaked in 60–90% ethyl or isopropyl alcohol to remove all blood and organic material.

STEP 2: Completely disassemble the laparoscopic equipment: operating laparoscope or Laprocator™, trocar, uterine manipulator, cervical vulsellum forceps, Verres needle and Falope Ring® guide kit.

STEP 3: Place disassembled parts in a basin of clean water and mild, nonabrasive soap.

STEP 4: Wash all outer surfaces, using a soft cotton cloth.

STEP 5: Clean inner channels with a cleaning brush supplied with the laparoscope kit. Use a circular motion to remove particulate matter. (Organic matter hidden in the narrow channels may cause infection later.) Be careful not to forcibly push the brush against the closed end of the inner tube as this may damage it.

STEP 6: Rinse all parts thoroughly with clean water (running water or from a basin) three times. Use the brush to remove soap and particles from the inner channels. (Soap, if not thoroughly rinsed away, will decrease the effectiveness of the disinfectant.)

STEP 7: Dry equipment with a clean soft cotton cloth or air dry. (Excess water will dilute the disinfectant, decreasing its effectiveness.)

STEP 8: Clean lenses at least weekly, and more often as needed, but do not touch the lenses with fingers (see **STEP 3**, below).

STEP 9: High-level disinfect (for 20 minutes) or sterilize (overnight), or if not needed immediately, carefully store in instrument container after cleaning

[1] *Adapted from*: Altobelli 1980.

and drying until next use. (Instruments should be high-level disinfected **immediately prior to use** to prevent recontamination.)

HOW TO CLEAN LENSES ON LAPAROSCOPES

STEP 1: Remove the plastic eyepiece of the laparoscope prior to cleaning the proximal lens with acetone or 60–90% alcohol. (Acetone and other organic solvents can severely damage plastic.)

STEP 2: Clean lenses with a **cotton swab** soaked in alcohol or acetone. (Cotton will not scratch the lens, and alcohol and acetone will not weaken the cement around the lens.)

STEP 3: While cleaning, do not touch lenses with fingers. (Skin oils may damage the lenses.)

STEP 4: Clean lenses at least weekly, and more often as needed.

HOW TO STERILIZE LAPAROSCOPES

STEP 1: Decontaminate, wash and dry all instruments to be sterilized as described above.

STEP 2: In a well-ventilated area, wearing gloves to prevent skin irritation, **completely immerse** clean, dry, disassembled instruments and cleaning brush in a plastic container at least 8 cm (3 inches) in depth that contains either 8% formaldehyde or a 2–4% glutaraldehyde (e.g., Cidex®). The disinfectant must touch all surfaces in order to be effective. (See **Appendix F** for directions on how to prepare and use these disinfectants.)

Note: Avoid placing instruments on top of each other, as this may damage them.

STEP 3: Cover the container during the disinfection procedure. (This will decrease the rate of evaporation and will keep dust out of the solution.)

STEP 4: Allow to soak **8 to 10 hours** in most glutaraldehydes, and **at least 24 hours** in 8% formaldehyde. Both agents work best at room temperature. Sterilization cannot be assured at temperatures less than 20°C (68°F). Because instructions vary, carefully read manufacturer's instructions for each product.

STEP 5: Use sterile gloves to carefully remove instruments from the solution. (Forceps or lifters may damage the instruments.)

STEP 6: Rinse **three times** with sterile water to completely remove all traces of the disinfectant. If sterile water is unavailable, rinse in cooled water which has been filtered and boiled for 20 minutes. Use a sterilized or high-level disinfected brush to assist with rinsing the narrow channels of the instruments. (This keeps movable parts from sticking due to any remaining disinfectant.) Finally, rinse completely with 60–90% ethyl or isopropyl alcohol. Allow to dry and use immediately. Do not store laparoscopes that have been rinsed with alcohol because residue can cause movable parts to stick.

STEP 7: Air dry in a sterile container with a cover. (Laparoscopes and accessories can be stored for up to 1 week in this container.)

HOW TO HIGH-LEVEL DISINFECT LAPAROSCOPES

STEP 1: Decontaminate, wash and dry all items to be high-level disinfected.

STEP 2: In a well-ventilated area, wearing gloves to prevent skin irritation, **completely immerse** clean, dry disassembled instruments and cleaning brush in a plastic container (as above) containing either 8% formaldehyde or a 2–4% glutaraldehyde (e.g. Cidex) solution. The disinfectant must touch all surfaces in order to be effective. (See **Appendix F** for directions on how to prepare and use these disinfectants.)

Note: Avoid placing instruments on top of each other, as this may damage them.

STEP 3: Cover the container during the HLD process. (This will decrease the rate of evaporation and will keep dust out of the solution.)

STEP 4: Allow to soak for **20 minutes**.

STEP 5: After 20 minutes, use high-level disinfected or sterile gloves to carefully remove instruments from the solution. (Forceps or lifters may damage the instruments.)

STEP 6: Rinse **three times** with cooled water that has been filtered and boiled for 20 minutes in order to completely remove all traces of the disinfectant. (This will prevent the solution from irritating the client's skin and keep the movable parts from sticking.) Although not necessary, sterile water can be used in place of boiled water. Use a high-level disinfected brush to assist with rinsing the narrow channels of the instruments.

STEP 7: Allow to air dry in a high-level disinfected container or dry with a high-level disinfected soft cotton cloth and place immediately on the instrument table.

HOW TO STORE LAPAROSCOPES

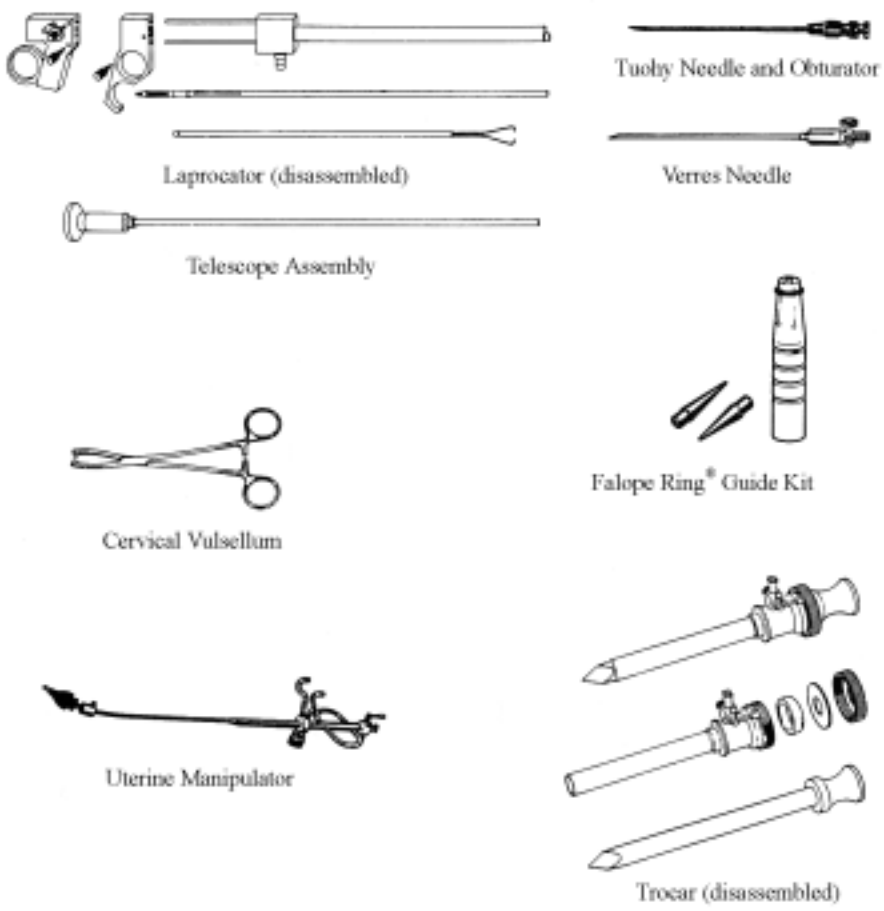
STEP 1: Decontaminate, wash and dry all instruments to be stored.

Remember: Before using stored laparoscopes and accessories such as trocars, they must be disassembled, cleaned and either sterilized or high-level disinfected.

STEP 2: Assemble laparoscope and trocar.

STEP 3: Place laparoscope and trocar in the padded container supplied with the equipment and store in a cool, dry place.

Figure H-1. Atlas of Laprocator System



Source: Altobelli 1980.

TIPS FOR PROLONGING THE LIFE OF LAPAROSCOPES[2]

- Failure to completely disassemble and clean the endoscope properly is the most common cause of problems. In addition, blood and other organic material left to dry on the instruments are difficult to remove and may be a source of infection.
- Never autoclave or boil laparoscopes because heat will damage the optics. Always **sterilize** or **high-level disinfect** with chemical sterilants or disinfectants such as glutaraldehyde or formaldehyde.
- Remove instruments from the disinfectant solution as soon as timing requirements are met. Prolonged immersion may shorten the life of the instrument.
- **Rinse at least three times** with cooled sterile (or boiled) water after cold sterilization or high-level disinfection respectively, to remove residue. Residue can cause movable parts to stick.

[2] *Adapted from*: Wolf R. 1984.

- Wear sterile or high-level disinfected gloves to handle instruments after final processing. Forceps and clamps may damage the laparoscope.
- Avoid picking up or handling instruments in groups or bunches.
- Always grasp the laprocator at the eyepiece end to avoid damaging the operative forceps.
- Avoid piling instruments or cables on top of each other to prevent damage or fiber breakage.
- Do not use Savlon® as it is not a high-level disinfectant and has been associated with clouding laparoscope optical lenses.

REFERENCES

Altobelli LC et al. 1980. *Laprocator™ Preventive Care and Maintenance*. JHPIEGO Corporation: Baltimore, MD.

Wolf R. 1984. *Instruction Manual: Gynecological Laparoscopy Instruments*. (Ref. E1-05-82). Richard Wolf Medical Instruments Corp.: Rosemont, IL.

APPENDIX I

TYPE AND DURATION OF PRECAUTIONS NEEDED FOR SELECTED INFECTIONS AND CONDITIONS[1]

Infection/Condition	Precautions	
	Type*	Duration†
Abscess		
Draining, major [a]	C	DI
Draining, minor or limited [b]	S	
Acquired immunodeficiency syndrome [c]	S	
Actinomycosis	S	
Adenovirus infection, in infants and young children	D,C	DI
Amebiasis	S	
Anthrax		
Cutaneous	S	
Pulmonary	S	
Antibiotic-associated colitis (see *Clostridium difficile*)		
Arthropodborne viral encephalitides (eastern, western, Venezuelan equine encephalomyelitis; St Louis, California encephalitis)	S [d]	
Arthropodborne viral fevers (dengue, yellow fever, Colorado tick fever)	S [d]	
Ascariasis	S	
Aspergillosis	S	
Babesiosis	S	
Blastomycosis, North American, cutaneous or pulmonary	S	
Botulism	S	
Bronchiolitis (see respiratory infections in infants and young children)		
Brucellosis (undulant, Malta, Mediterranean fever)	S	
Campylobacter gastroenteritis (see gastroenteritis)		
Candidiasis, all forms including mucocutaneous	S	
Cat-scratch fever (benign inoculation lymphoreticulosis)	S	
Cellulitis, uncontrolled drainage	C	DI
Chancroid (soft chancre)	S	
Chickenpox (varicella; see F [e] for varicella exposure)	A,C	F [e]
Chlamydia trachomatis		
Conjunctivitis	S	
Genital	S	
Respiratory	S	
Cholera (see gastroenteritis)		
Closed-cavity infection		
Draining, limited or minor	S	
Not draining	S	
Clostridium		
C botulinum	S	
C difficile	C	DI

[1] *Source*: Garner JS and HICPAC 1996.

Infection/Condition	Precautions	
	Type[*]	Duration[†]
C perfringens		
Food poisoning	S	
Gas gangrene	S	
Coccidioidomycosis (valley fever)		
Draining lesions	S	
Pneumonia	S	
Colorado tick fever	S	
Congenital rubella	C	F [f]
Conjunctivitis		
Acute bacterial	S	
Chlamydia	S	
Gonococcal	S	
Acute viral (acute hemorrhagic)	C	DI
Coxsackievirus disease (see enteroviral infection)		
Creutzfeldt-Jakob disease	S [g]	
Croup (see respiratory infections in infants and young children)		
Cryptococcosis	S	
Cryptosporidiosis (see gastroenteritis)		
Cysticercosis	S	
Cytomegalovirus infection, neonatal or immunosuppressed	S	
Decubitus ulcer, infected		
Major [a]	C	DI
Minor or limited [b]	S	
Dengue	S [d]	
Diarrhea, acute-infective etiology suspected (see gastroenteritis)		
Diphtheria		
Cutaneous	C	CN [h]
Pharyngeal	D	CN [h]
Ebola viral hemorrhagic fever	C [i]	DI
Echinococcosis (hydatidosis)	S	
Echovirus (see enteroviral infection)		
Encephalitis or encephalomyelitis (see specific etiologic agents)		
Endometritis	S	
Enterobiasis (pinworm disease, oxyuriasis)	S	
Enterococcus species (see multidrug-resistant organisms if epidemiologically significant or vancomycin-resistant)		
Enterocolitis, *Clostridium difficile*	C	DI
Enteroviral infections		
Adults	S	
Infants and young children	C	DI
Epiglottitis, due to *Haemophilus influenzae*	D	U(24 hrs)
Epstein-Barr virus infection, including infectious mononucleosis	S	
Erythema infectiosum (also see Parvovirus B19)	S	
Escherichia coli gastroenteritis (see gastroenteritis)		
Food poisoning		
Botulism	S	
Clostridium perfringens or welchii	S	
Staphylococcal	S	

Infection/Condition	Precautions	
	Type[*]	Duration[†]
Furunculosis-staphylococcal		
Infants and young children	C	DI
Gangrene (gas gangrene)	S	
Gastroenteritis		
Campylobacter species	S [j]	
Cholera	S [j]	
Clostridium difficile	C	DI
Cryptosporidium species	S [j]	
Escherichia coli		
Enterohemorrhagic O157:H7	S [j]	
Diapered or incontinent	C	DI
Other species	S [j]	
Giardia lamblia	S [j]	
Rotavirus	S [j]	
Diapered or incontinent	C	DI
Salmonella species (including *S typhi*)	S [j]	
Shigella species	S [j]	
Diapered or incontinent	C	DI
Vibrio parahaemolyticus	S [j]	
Viral (if not covered elsewhere)	S [j]	
Yersinia enterocolitica	S [j]	
German measles (see rubella)		
Giardiasis (see gastroenteritis)		
Gonococcal ophthalmia neonatorum (gonorrheal ophthalmia, acute conjunctivitis of newborn)	S	
Gonorrhea	S	
Granuloma inguinale (donovanosis, granuloma venereum)	S	
Guillain-Barré, syndrome	S	
Hand, foot, and mouth disease (see enteroviral infection)		
Hantavirus pulmonary syndrome	S	
Helicobacter pylori	S	
Hemorrhagic fevers (for example, Lassa and Ebola)	C [i]	DI
Hepatitis, viral		
Type A	S	
Diapered or incontinent patients	C	F [k]
Type B-HBsAg positive	S	
Type C and other unspecified non-A, non-B	S	
Type E	S	
Herpangina (see enteroviral infection)		
Herpes simplex (*Herpesvirus hominis*)		
Encephalitis	S	
Neonatal [l] (see F [l] for neonatal exposure)	C	DI
Mucocutaneous, disseminated or primary, severe	C	DI
Mucocutaneous, recurrent (skin, oral, genital)	S	
Herpes zoster (varicella-zoster)		
Localized in immunocompromised patient, or disseminated	A,C	DI [m]
Localized in normal patient	S [m]	
Histoplasmosis	S	

Infection/Condition	Precautions	
	Type[*]	Duration[†]
HIV (see human immunodeficiency virus)	S	
Hookworm disease (ancylostomiasis, uncinariasis)	S	
Human immunodeficiency virus (HIV) infection [c]	S	
Impetigo	C	U (24 hrs)
Infectious mononucleosis	S	
Influenza	D [n]	DI
Kawasaki syndrome	S	
Lassa fever	C [i]	DI
Legionnaires' disease	S	
Leprosy	S	
Leptospirosis	S	
Lice (pediculosis)	C	U (24 hrs)
Listeriosis	S	
Lyme disease	S	
Lymphocytic choriomeningitis	S	
Lymphogranuloma venereum	S	
Malaria	S [d]	
Marburg virus disease	C [i]	DI
Measles (rubeola), all presentations	A	DI
Melioidosis, all forms	S	
Meningitis		
Aseptic (nonbacterial or viral meningitis; also see enteroviral infections)	S	
Bacterial, gram-negative enteric, in neonates	S	
Fungal	S	
Haemophilus influenzae, known or suspected	D	U(24 hrs)
Listeria monocytogenes	S	
Neisseria meningitidis (meningococcal) known or suspected	D	U(24 hrs)
Pneumococcal	S	
Tuberculosis [o]	S	
Other diagnosed bacterial	S	
Meningococcal pneumonia	D	U(24 hrs)
Meningococcemia (meningococcal sepsis)	D	U(24 hrs)
Molluscum contagiosum	S	
Mucormycosis	S	
Multidrug-resistant organisms, infection or colonization [p]		
Gastrointestinal	C	CN
Respiratory	C	CN
Pneumococcal	S	
Skin, wound, or burn	C	CN
Mumps (infectious parotitis)	D	F [q]
Mycobacteria, nontuberculosis (atypical)		
Pulmonary	S	
Wound	S	
Mycoplasma pneumonia	D	DI
Necrotizing enterocolitis	S	
Nocardiosis, draining lesions or other presentations	S	
Norwalk agent gastroenteritis (see viral gastroenteritis)		

Infection/Condition	Precautions	
	Type[*]	**Duration**[†]
Orf	S	
Parainfluenza virus infection, respiratory in infants and young children	C	DI
Parvovirus B19	D	F [r]
Pediculosis (lice)	C	U(24 hrs)
Pertussis (whooping cough)	D	F [s]
Pinworm infection	S	
Plague		
Bubonic	S	
Pneumonic	D	U(72 hrs)
Pleurodynia (see enteroviral infection)		
Pneumonia		
Adenovirus	D,C	DI
Bacterial not listed elsewhere (including gram-negative bacterial)	S	
Burkholderia cepacia in cystic fibrosis (CF) patients, including respiratory tract colonization	S [t]	
Chlamydia	S	
Fungal	S	
Haemophilus influenzae		
Adults	S	
Infants and children (any age)	D	U(24 hrs)
Legionella	S	
Meningococcal	D	U(24 hrs)
Multidrug-resistant bacterial (see multidrug-resistant organisms)		
Mycoplasma (primary atypical pneumonia)	D	DI
Pneumococcal	S	
Multidrug-resistant (see multidrug-resistant organisms)		
Pneumocystis carinii	S [u]	
Pseudomonas cepacia (see *Burkholderia cepacia*)	S [t]	
Staphylococcus aureus	S	
Streptococcus, group A		
Adults	S	
Infants and young children	D	U(24hrs)
Viral		
Adults	S	
Infants and young children (see respiratory infectious disease, acute)		
Poliomyelitis	S	
Psittacosis (ornithosis)	S	
Q fever	S	
Rabies	S	
Rat-bite fever (*Streptobacillus moniliformis* disease, *Spirillum minus* disease)	S	
Relapsing fever	S	
Resistant bacterial infection or colonization (see multidrug-resistant organisms)		
Respiratory infectious disease, acute (if not covered elsewhere)		
Adults	S	
Infants and young children [c]	C	DI
Respiratory syncytial virus infection, in infants and young children, and immunocompromised adults	C	DI

Infection/Condition	Precautions	
	Type[*]	Duration[†]
Reye's syndrome	S	
Rheumatic fever	S	
Rickettsial fevers, tickborne (Rocky Mountain spotted fever, tickborne typhus fever)	S	
Rickettsialpox (vesicular rickettsiosis)	S	
Ringworm (dermatophytosis, dermatomycosis, tinea)	S	
Ritter's disease (staphylococcal scalded skin syndrome)	S	
Rocky Mountain spotted fever	S	
Roseola infantum (exanthem subitum)	S	
Rotavirus infection (see gastroenteritis)		
Rubella (German measles; also see congenital rubella)	D	F [v]
Salmonellosis (see gastroenteritis)		
Scabies	C	U(24 hrs)
Scalded skin syndrome, staphylococcal (Ritter's disease)	S	
Schistosomiasis (bilharziasis)	S	
Shigellosis (see gastroenteritis)		
Sporotrichosis	S	
Spirillum minus disease (rat-bite fever)	S	
Staphylococcal disease (*S aureus*)		
Skin, wound, or burn		
Major [a]	C	DI
Minor or limited [b]	S	
Enterocolitis	S [j]	
Multidrug-resistant (see multidrug-resistant organisms)		
Pneumonia	S	
Scalded skin syndrome	S	
Toxic shock syndrome	S	
Streptobacillus moniliformis disease (rat-bite fever)	S	
Streptococcal disease (group A streptococcus)		
Skin, wound, or burn		
Major [a]	C	U(24 hrs)
Minor or limited [b]	S	
Endometritis (puerperal sepsis)	S	
Pharyngitis in infants and young children	D	U(24 hrs)
Pneumonia in infants and young children	D	U(24 hrs)
Scarlet fever in infants and young children	D	U(24 hrs)
Streptococcal disease (group B streptococcus), neonatal	S	
Streptococcal disease (not group A or B) unless covered elsewhere	S	
Multidrug-resistant (see multidrug-resistant organisms)		
Strongyloidiasis	S	
Syphilis		
Skin and mucous membrane, including congenital, primary, secondary	S	
Latent (tertiary) and seropositivity without lesions	S	
Tapeworm disease		
Hymenolepis nana	S	
Taenia solium (pork)	S	
Other	S	
Tetanus	S	

Infection/Condition	Precautions	
	Type[*]	**Duration**[†]
Tinea (fungus infection dermatophytosis, dermatomycosis, ringworm)	S	
Toxoplasmosis	S	
Toxic shock syndrome (staphylococcal disease)	S	
Trachoma, acute	S	
Trench mouth (Vincent's angina)	S	
Trichinosis	S	
Trichomoniasis	S	
Trichuriasis (whipworm disease)	S	
Tuberculosis		
Extrapulmonary, draining lesion (including scrofula)	S	
Extrapulmonary, meningitis [o]	S	
Pulmonary, confirmed or suspected or laryngeal disease	A	F [w]
Skin-test positive with no evidence of current pulmonary disease	S	
Tularemia		
Draining lesion	S	
Pulmonary	S	
Typhoid (*Salmonella typhi*) fever (see gastroenteritis)		
Typhus, endemic and epidemic	S	
Urinary tract infection (including pyelonephritis), with or without urinary catheter	S	
Varicella (chickenpox)	A,C	F [e]
Vibrio parahaemolyticus (see gastroenteritis)		
Vincent's angina (trench mouth)	S	
Viral diseases		
Respiratory (if not covered elsewhere)		
Adults	S	
Infants and young children (see respiratory infectious disease, acute)		
Whooping cough (pertussis)	D	F [s]
Wound infections		
Major [a]	C	DI
Minor or limited [b]	S	
Yersinia enterocolitica gastroenteritis (see gastroenteritis)		
Zoster (varicella-zoster)		
Localized in immunocompromised patient, disseminated	A,C	DI [m]
Localized in normal patient	S [m]	
Zygomycosis (phycomycosis, mucormycosis)	S	

Abbreviations:

* Type of Precautions: A, Airborne; C, Contact; D, Droplet; S, Standard; when A, C, and D are specified, also use S.
† Duration of precautions: CN, until off antibiotics and culture-negative; DI, duration of illness (with wound lesions, DI means until they stop draining); U, until time specified in hours (hrs) after initiation of effective therapy; F, see footnote.
[a] No dressing or dressing does not contain drainage adequately.
[b] Dressing covers and contains drainage adequately.
[c] Also see syndromes or conditions listed in Table 2.
[d] Install screens in windows and doors in endemic areas.
[e] Maintain precautions until all lesions are crusted. The average incubation period for varicella is 10 to 16 days, with a range of 10 to 21 days. After exposure, use varicella zoster immune globulin (VZIG) when appropriate, and discharge susceptible patients if possible. Place exposed susceptible patients on Airborne Precautions beginning 10 days after exposure and continuing until 21 days after last exposure (up to 28 days if VZIG has been given). Susceptible persons should not enter the room of patients on precautions if other immune caregivers are available.
[f] Place infant on precautions during any admission until 1 year of age, unless nasopharyngeal and urine cultures are negative for virus after age 3 months.
[g] Additional special precautions are necessary for handling and decontamination of blood, body fluids and tissues, and contaminated items from patients with confirmed or suspected disease. See latest College of American Pathologists (Northfield, Illinois) guidelines or other references.
[h] Until two cultures taken at least 24 hours apart are negative.
[i] Call state health department and CDC for specific advice about management of a suspected case. During the 1995 Ebola outbreak in Zaire, interim recommendations were published. (97) Pending a comprehensive review of the epidemiologic data from the outbreak and evaluation of the interim recommendations, the 1988 guidelines for management of patients with suspected viral hemorrhagic infections (16) will be reviewed and updated if indicated.
[j] Use Contact Precautions for diapered or incontinent children <6 years of age for duration of illness.
[k] Maintain precautions in infants and children <3 years of age for duration of hospitalization; in children 3 to 14 years of age, until 2 weeks after onset of symptoms; and in others, until 1 week after onset of symptoms.
[l] For infants delivered vaginally or by C-section and if mother has active infection and membranes have been ruptured for more than 4 to 6 hours.
[m] Persons susceptible to varicella are also at risk for developing varicella when exposed to patients with herpes zoster lesions; therefore, susceptibles should not enter the room if other immune caregivers are available.
[n] The "Guideline for Prevention of Nosocomial Pneumonia" (95,96) recommends surveillance, vaccination, antiviral agents, and use of private rooms with negative air pressure as much as feasible for patients for whom influenza is suspected or diagnosed. Many hospitals encounter logistic difficulties and physical plant limitations when admitting multiple patients with suspected influenza during community outbreaks. If sufficient private rooms are unavailable, consider cohorting patients or, at the very least, avoid room sharing with high-risk patients. See “Guideline for Prevention of Nosocomial Pneumonia” (95,96) for additional prevention and control strategies.
[o] Patient should be examined for evidence of current (active) pulmonary tuberculosis. If evidence exists, additional precautions are necessary (see tuberculosis).
[p] Resistant bacteria judged by the infection control program, based on current state, regional, or national recommendations, to be of special clinical and epidemiologic significance.
[q] For 9 days after onset of swelling.
[r] Maintain precautions for duration of hospitalization when chronic disease occurs in an immunodeficient patient. For patients with transient aplastic crisis or red-cell crisis, maintain precautions for 7 days.
[s] Maintain precautions until 5 days after patient is placed on effective therapy.
[t] Avoid cohorting or placement in the same room with a CF patient who is not infected or colonized with *B cepacia*. Persons with CF who visit or provide care and are not infected or colonized with *B cepacia* may elect to wear a mask when within 3 ft of a colonized or infected patient.
[u] Avoid placement in the same room with an immunocompromised patient.
[v] Until 7 days after onset of rash.
[w] Discontinue precautions *only* when TB patient is on effective therapy, is improving clinically, and has three consecutive negative sputum smears collected on different days, or TB is ruled out. Also see CDC “Guidelines for Preventing the Transmission of Tuberculosis in Health-Care Facilities.”(23)

REFERENCES

Garner JS and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53–80, and *Am J Infect Control* 24(1): 24–52.

APPENDIX J

CDC RECOMMENDATIONS FOR PREVENTION OF SURGICAL SITE INFECTION[1]

RATIONALE

The Guidelines for Prevention of Surgical Site Infection (1999) provides recommendations concerning reduction of surgical site infection (SSI) risk. Each recommendation is categorized on the basis of existing scientific data, theoretical rationale, and applicability.

Category I recommendations, including IA and IB, are those recommendations that are viewed as effective by HICPAC and experts in the fields of surgery, infectious diseases and infection control. Both Category IA and IB recommendations are applicable for, and should be adopted by, all healthcare facilities; IA and IB recommendations differ only in the strength of the supporting scientific evidence.

Category II recommendations are supported by less scientific data than Category I recommendations; such recommendations may be appropriate for addressing specific nosocomial problems or specific patient populations.

No recommendation is offered for some practices, either because there is a lack of consensus regarding their efficacy or because the available scientific evidence is insufficient to support their adoption. For such unresolved issues, practitioners should use judgment to determine a policy regarding these practices within their organization.

RANKING

Category IA. Strongly recommended for implementation and supported by well-designed experimental, clinical or epidemiological studies.

Category IB. Strongly recommended for implementation and supported by some experimental, clinical or epidemiological studies and strong theoretical rationale.

Category II. Suggested for implementation and supported by suggestive clinical or epidemiological studies or theoretical rationale.

No recommendation (unresolved issue). Practices for which insufficient evidence or no consensus regarding efficacy exists.

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[1] *Adapted from*: Mangram, HICPAC and CDC 1999.

RECOMMENDATIONS

1. PREOPERATIVE

a. Preparation of the patient

1. Whenever possible, identify and treat all infections remote to the surgical site before elective operation and postpone elective operations on patients with remote site infections until the infection has resolved. *Category IA*
2. Do not remove hair preoperatively unless the hair at or around the incision site will interfere with the operation. *Category IA*
3. If hair is removed, remove immediately before the operation, preferably with electric clippers. *Category IA*
4. Adequately control serum blood glucose levels in all diabetic patients and particularly avoid hyperglycemia perioperatively. *Category IB*
5. Encourage stopping use of tobacco products. At minimum, instruct patients to abstain for at least 30 days before elective operation from smoking cigarettes, cigars, pipes or any other form of tobacco consumption (e.g., chewing/dipping). *Category IB*
6. Do not withhold necessary blood products from surgical patients as a means to prevent SSI. *Category IB*
7. Require patients to shower or bathe with an antiseptic agent on at least the night before the operative day. *Category IB*
8. Thoroughly wash and clean at and around the incision site to remove gross contamination before performing antiseptic skin preparation. *Category IB*
9. Use an appropriate antiseptic agent for skin preparation (Table 6). *Category IB*
10. Apply preoperative antiseptic skin preparation in concentric circles moving toward the periphery. The prepared area must be large enough to extend the incision or create new incisions or drain sites, if necessary. *Category II*
11. Keep preoperative hospital stay as short as possible while allowing for adequate preoperative preparation of the patient. *Category II*
12. No recommendation to taper or discontinue systemic steroid use (when medically permissible) before elective operation. *Unresolved issue*
13. No recommendation to enhance nutritional support for surgical patients solely as a means to prevent SSI. *Unresolved issue*
14. No recommendation to preoperatively apply mupirocin to the nose (nares) to prevent SSI. *Unresolved issue*

b. Hand/forearm antisepsis for surgical team members

1. Keep nails short and do not wear artificial nails. *Category IB*

2. Perform a preoperative surgical scrub for at least 2 to 5 minutes using an appropriate antiseptic. Scrub the hands and forearms up to the elbows. *Category IB*
3. After performing the surgical scrub, keep hands up and away from the body (elbows in flexed position) so that water runs from the tips of the fingers toward the elbows. Dry hands with a sterile towel and put on a sterile gown and gloves. *Category IB*
4. Clean underneath each fingernail prior to performing the first surgical scrub of the day. *Category II*
5. Do not wear hand or arm jewelry. *Category II*
6. No recommendation on wearing nail polish. *Unresolved Issue*

c. Management of infected or colonized surgical personnel

1. Educate and encourage surgical personnel who have signs and symptoms of a transmissible infectious illness to report conditions promptly to their supervisory and occupational health service personnel. *Category IB*
2. Develop well-defined policies concerning patient care responsibilities when personnel have potentially transmissible infectious conditions. These policies should govern: (a) personnel responsibility in using the health service and reporting illness, (b) work restrictions, and (c) clearance to resume work after an illness that required work restriction. The policies also should identify persons who have the authority to remove personnel from duty. *Category IB*
3. Exclude from duty surgical personnel who have draining skin lesions until infection has been ruled out or personnel have received adequate therapy and infection has resolved. *Category IB*
4. Do not routinely exclude surgical personnel who are colonized with organisms such as *S. aureus* (nose, hands or other body site) or group A *Streptococcus*, unless such personnel have been linked epidemiologically to dissemination of the organism in the healthcare setting. *Category IB*

d. Antimicrobial prophylaxis

1. Administer a prophylactic antimicrobial agent only when indicated, and select it based on its efficacy against the most common pathogens causing SSI for a specific operation and published recommendations. *Category IA*
2. Administer by the intravenous route the initial dose of prophylactic antimicrobial agent, timed such that a bactericidal concentration of the drug is established in serum and tissues when the incision is made. Maintain therapeutic levels of the agent in serum and tissues throughout the operation and until, at most, a few hours after the incision is closed in the operating room. *Category IA*
3. Before elective colorectal operations in addition to d2 above, mechanically prepare the colon by use of enemas and cathartic

agents. Administer nonabsorbable oral antimicrobial agents in divided doses on the day before the operation. *Category IA*

4. For high-risk cesarean section, administer the prophylactic antimicrobial agent immediately after the umbilical cord is clamped. *Category IA*
5. Do not routinely use vancomycin for antimicrobial prophylaxis. *Category IB*

2. INTRAOPERATIVE

a. Ventilation

1. Maintain positive-pressure ventilation in the operating room with respect to the corridors and adjacent areas. *Category IB*
2. Maintain a minimum of 15 air changes per hour, of which at least 3 should be fresh air. *Category IB*
3. Filter all air, recirculated and fresh, through the appropriate filters per the American Institute of Architects' recommendations. *Category IB*
4. Introduce all air at the ceiling, and exhaust near the floor. *Category IB*
5. Do not use UV radiation in the operating room to prevent SSI. *Category IB*
6. Keep operating room doors closed except as needed for passage of equipment, personnel and the patient. *Category IB*
7. Consider performing orthopedic implant operations in operating rooms supplied with ultraclean air. *Category II*
8. Limit the number of personnel entering the operating room to necessary personnel. *Category II*

b. Cleaning and disinfection of environmental surfaces

1. When visible soiling or contamination with blood or other body fluids of surfaces or equipment occurs during an operation, use disinfectant to clean the affected areas before the next operation. *Category IB*
2. Do not perform special cleaning or closing of operating rooms after contaminated or dirty operations. *Category IB*
3. Do not use tacky mats at the entrance to the operating room suite or individual operating rooms for infection control. *Category IB*
4. Wet vacuum the operating room floor after the last operation of the day or night with disinfectant. *Category II*
5. No recommendation on disinfecting environmental surfaces or equipment used in operating rooms between operations in the absence of visible soiling. *Unresolved issue*

c. Microbiologic sampling

1. Do not perform routine environmental sampling of the operating room. Perform microbiologic sampling of operating room environmental surfaces or air only as part of an epidemiologic investigation. *Category IB*

d. Sterilization of surgical instruments

1. Sterilize all surgical instruments according to published guidelines. *Category IB*
2. Perform flash sterilization only for patient care items that will be used immediately (e.g., to reprocess an inadvertently dropped instrument). Do not use flash sterilization for reasons of convenience, as an alternative to purchasing additional instrument sets, or to save time. *Category IB*

e. Surgical attire and drapes

1. Wear a surgical mask that fully covers the mouth and nose when entering the operating room if an operation is about to begin or already under way or if sterile instruments are exposed. Wear the mask throughout the operation. *Category IB*
2. Wear a cap or hood to fully cover hair on the head and face when entering the operating room. *Category IB*
3. Do not wear shoe covers for the prevention of SSI. *Category IB*
4. Wear sterile gloves if a scrubbed surgical team member. Put on gloves after putting on a sterile gown. *Category IB*
5. Use surgical gowns and drapes that are effective barriers when wet (i.e., materials that resist liquid penetration). *Category IB*
6. Change scrub suits that are visibly soiled, contaminated and/or penetrated by blood or other potentially infectious materials. *Category IB*
7. No recommendations on how or where to launder scrub suits, on restricting use of scrub suits to the operating suite or for covering scrub suits when out of the operating suite. *Unresolved issue*

f. Asepsis and surgical technique

1. Adhere to principles of asepsis when placing intravascular devices (e.g., central venous catheters), spinal or epidural anesthesia catheters, or when dispensing and administering intravenous drugs. *Category IA*
2. Assemble sterile equipment and solutions immediately prior to use. *Category II*
3. Handle tissue gently, maintain effective hemostasis, minimize devitalized tissue and foreign bodies (i.e., sutures, charred tissues, necrotic debris) and eradicate dead space at the surgical site. *Category IB*
4. Use delayed primary skin closure or leave an incision open to heal by second intention if the surgeon considers the surgical site to be heavily contaminated (e.g., Class III and Class IV). *Category IB*
5. If drainage is necessary, use a closed suction drain. Place a drain through a separate incision distant from the operative incision. Remove the drain as soon as possible. *Category IB*

3. POSTOPERATIVE INCISION CARE

a. Protect with a sterile dressing for 24 to 48 hours postoperatively an incision that has been closed primarily. *Category IB*
b. Wash hands before and after dressing changes and any contact with the surgical site. *Category IB*
c. When an incision dressing must be changed, use sterile technique. *Category II*
d. Educate the patient and family regarding proper incision care, symptoms of SSI, and the need to report such symptoms. *Category II*
e. No recommendation to cover an incision closed primarily beyond 48 hours, nor on the appropriate time to shower or bathe with an uncovered incision. *Unresolved issue*

REFERENCES

Mangram AJ, Hospital Infection Control Practices Advisory Committee (HICPAC) and Centers for Disease Control and Prevention (CDC). 1999. Guidelines for prevention of surgical site infection. *Infect Control Hosp Epidemiol* 24(4): 247–278.
Available at: http://www.cdc.gov/ncidod/hip/SSI/SSI_guideline.htm

APPENDIX K

FETAL AND NEWBORN INFECTIOUS DISEASE PREVENTION[1]

BACTERIAL INFECTIONS

Group B Streptococcal Septicemia

Since its emergence in the 1970s, group B streptococcal sepsis has been the leading bacterial infection associated with illness and death in newborns in developed countries. For infants with the infection, the mortality rate is up to 25% even with early diagnosis and prompt treatment. Colonization of the vagina and rectum in pregnancy is common (from 10–40% of women during late pregnancy). Although colonization is usually without symptoms, this pathogen causes considerable maternal and fetal infection before and after delivery (e.g., chorioamnionitis and septicemia of the fetus and newborn, and urinary tract infections, endometritis and wound infections in postpartum women).

About 50% of newborn infants exposed during delivery to mothers colonized with this pathogen will become colonized. Fortunately, maternally transmitted antibodies protect most of these infants. Thus, only a few (1–2 per 1000 live births) develop clinical disease. Preterm or low-birth weight infants are at the highest risk (about 25%) of developing this serious, often fatal, infection during the first week of life. This is due, in part, to the reduced ability of a preterm infant's immune system to resist infection.

Prophylaxis

Where antenatal services include laboratory testing, most neonatal group B streptococcal infections can be prevented through the use of intrapartum antimicrobial prophylaxis in women at increased risk of transmitting the infection to their newborns (CDC 2000; Keenan 1998). Such women can be identified by having a positive anogenital culture for this pathogen at 35–37 weeks or at least one of the following risk factors associated with early-onset infections:

- group B streptococci bacteriuria during pregnancy;
- previously delivered infant infected with group B streptococcal infection;
- preterm birth (before 37 weeks gestation);
- rupture of membranes (>18 hours); and
- clinically evident intra-amniotic infection syndrome (chorioamnionitis) with maternal temperature greater than 38°C (100.4°F), or prior infected child.

[1] *Adapted from*: AAP and ACOG 1997.

Prophylactic antibiotic treatment, as outlined in **Table K-1,** should begin as soon as labor starts or a risk factor is identified.

Table K-1. Prophylaxis of Group B Streptococcal Infection

PENICILLIN ALLERGY: MATERNAL STATUS	RECOMMENDED TREATMENT	ALTERNATIVE TREATMENT
No known allergy	Penicillin G: 5 million units given in an IV load; then 2.5 million units every 4 hours until delivery	Ampicillin: 2 g given in an IV load; then 1g IV every 4 hours until delivery
Allergy	Clindamycin (Cleocin): 900 mg IV every 8 hours until delivery	Erythromycin: 500 mg IV every 6 hours until delivery

IV= intravenous

Adapted from: CDC 1996.

Prevention measures in the nursery or neonatal intensive care unit (NICU) for infants at risk of group B streptococcal disease, who are being treated prophylactically, should stress hand hygiene, use of gloves and avoidance of crowding because cross-contamination (infant to infant) can occur on the hands of health workers. Newborns with early onset group B streptococcal disease can be treated in the NICU provided Standard Precautions, including Transmission-Based (Contact) Precautions, are used. (See **Chapters 2** and **21** for details.)

Chlamydial Infection

In many countries, 20–40% of pregnant women are infected with *Chlamydia trachomatis,* a sexually transmitted infection that is common in women who are young (age less than 24) and have multiple sex partners. Chlamydia is transmitted to newborns from infected mothers during birth, and 60–70% of infants delivered vaginally from infected mothers will acquire this infection. Of those infected newborns, 30–50% will develop purulent conjunctivitis unless treated prophylactically at birth with antibiotic eyedrops (tetracycline or erythromycin). Neonatal pneumonia occurs in another 10–20%.

Prevention during pregnancy includes treatment of infected pregnant women in the third trimester (after 30 weeks gestation) with erythromycin (tetracycline should not be used because it is deposited in the teeth of the developing fetus). Because antenatal testing is not available in most developing countries, use of eye drops is the only preventive measure usually available. Unfortunately, neither tetracycline nor erythromycin eye drops prevent chlamydial pneumonia. Prophylactic cesarean section is not recommended because the pneumonia is usually mild and easily and inexpensively treated.

Newborns with purulent conjunctivitis can be kept in the nursery or NICU provided Standard Precautions, including Transmission-Based (Contact)

Precautions, are used. (See **Chapters 2** and **21** for details.) In addition, all waste items (gauze or cotton wet with drainage from the eyes) should be disposed of in a plastic bag or leakproof, covered waste container.

Gonorrheal Infection

Prevention during pregnancy includes treatment of infected pregnant women with an appropriate antibiotic (tetracycline should not be used because it is deposited in the teeth of the developing fetus). Because antenatal testing is not available in most developing countries, use of eye drops (tetracycline or erythromycin) is the only preventative measure usually available.

Newborns with purulent conjunctivitis can be kept in the nursery or NICU provided Standard Precautions, including Transmission-Based (Contact) Precautions, are used. (See **Chapters 2** and **21** for details.) In addition, all waste items (gauze or cotton wet with drainage from the eyes) should be disposed of in a plastic bag or leakproof, covered waste container.

Listeriosis

Infection of the fetus or newborn with *Listeria monocytogenes* can occur antenatally (transfer across the placenta), during labor and delivery (vertical transmission) and nosocomially though contact with infected mothers or health workers. Because antenatal testing is not available in most developing countries, use of eye drops (tetracycline or erythromycin) is the only preventative measure usually available.

The clinical findings in infants with listeriosis are not well defined and often nonspecific, but they may be similar to group B streptococcal disease with early or late onset syndromes. Prompt diagnosis in the nursery is important because the infection is easily treated with penicillin or ampicillin.

Newborns at risk or with active listeriosis can be kept in the nursery or NICU provided Standard Precautions, including Transmission-Based (Contact) Precautions, are used. (See **Chapters 2** and **21** for details.) In addition, for newborns with conjunctivitis, all waste items (gauze or cotton wet with drainage from the eyes) should be disposed of in a plastic bag or leakproof, covered waste container.

Neonatal Tetanus

Neonatal tetanus is a major health problem in many developing countries where maternity services are limited and immunization against tetanus is inadequate. Although in the past 5 years progress has been made in reducing deaths from neonatal tetanus, WHO estimates that more than 500,000 deaths still occur annually in developing countries. Most newborns with tetanus have been born to nonimmunized mothers who delivered at home.

Infants become infected during delivery through use of an unclean instrument (e.g., strips of bamboo) to cut the umbilical cord or following delivery by placing substances heavily contaminated with tetanus

endospores (e.g., ashes, cow dung or dust from the hearth or doorway to the house) on the umbilical stump. Often this is done as part of a traditional birthing practice.

Prevention of neonatal tetanus, which has a 60–80% case-fatality rate, can be achieved through a combination of:

- immunizing all women of childbearing age, especially pregnant women;
- improving the quality and availability of maternity care; and
- educating mothers, relatives and birth attendants of the need for cutting the cord with a clean instrument and keeping the cord stump clean.

To be effective, nonimmunized pregnant women should receive at least two doses of tetanus toxoid prior to delivery—the first at the initial clinic visit and the second 4 weeks after the first and preferably at least 2 weeks before delivery. If a woman receives at least three additional doses over the next couple of years, she will be protected for life.

Syphilis

Antenatal testing of pregnant women should be done to identify and treat those women seropositive for syphilis and to prevent congenital syphilis in their newborns. If the results of serologic tests for syphilis were equivocal or not available, a cord blood or venous sample from the newborn should be tested.

Although the clinical findings of congenital syphilis in the newborn may be nonspecific, moist open lesions, which may be due to syphilis, are infectious. Therefore, care of the newborn in the nursery or NICU should include use of Standard Precautions, including Transmission-Based (Contact) Precautions, to prevent spread to other infants and health workers through cross-contamination. (See **Chapters 2** and **21** for details). In addition, all dressings covering the lesions should be disposed of in a plastic bag or leakproof, covered waste container.

VIRAL INFECTIONS

Hepatitis B

Note: No special care of these infants is required other than removal of maternal blood from the planned injection site to avoid introducing some of the virus contaminating the skin.

In many countries, 20–50% of pregnant women are seropositive for HBV, but prenatal screening for HBV by testing for hepatitis B surface antigen (HbsAg) is not available even for "high-risk" women. (Historical information about risk factors identifies less than half of chronic carriers.) Where HBV is endemic all infants should receive HBV vaccine within 12 hours because the majority of infants born to HBV-infected mothers will become infected, and about 70–90% chronic carriers. (If available, hepatitis B immune globulin prophylaxis should be given to infants at the same time.) About 95% of infants will be protected when the three-dose immunization series with HBV vaccine is completed; therefore,

breastfeeding of immunized babies born to HbsAg-positive women poses no additional risk of transmission of HBV.

Note: After blood has been removed, gloves do not need to be worn for changing diapers and other routine nursing care.

After delivery, the baby should be wiped with cotton (not gauze) dipped in clean water to remove blood and amniotic fluid from the newborn skin. Doing this minimizes the risk of exposing other infants or healthcare staff to blood or potentially contaminated amniotic fluid. After use, dispose of the cotton in a plastic bag or leakproof, covered waste container. There is no need for special precautions or isolation of newborn with an HBV infected mother; therefore, infants born to infected mothers (whether or not they receive hepatitis B vaccine) can stay in the nursery or NICU. Hospital staff need only use Standard Precautions in order to prevent exposure to blood or potentially contaminated body fluids.

Hepatitis C Virus

There is no vaccine or treatment to prevent HCV in humans; therefore, at present, primary prevention needs to be vigorously promoted. This involves education combined with behavior change interventions aimed at promoting safer sexual practices. Also, because HCV, HBV and HIV are transmitted by the same mechanisms, behavior change messages should be linked to AIDS and hepatitis B prevention efforts as well.

After delivery, care of HCV infected mothers and infants at risk of infection is the same as for HBV. Because HCV is found in breastmilk, women who are seropositive for HCV should be counseled about the risks of breastfeeding.

Herpes Simplex Virus

Women with active genital lesions who delivery vaginally have a 50% risk of transmitting the infection to their newborn if the infection is primary, but only 0–8% risk if recurrent. Most (70%) of newborns infected with HSV, however, are delivered from women who have neither active genital herpes nor a history of infection. Where possible, pregnant women at term (37 weeks or more) with documented genital lesions and intact membranes should be delivered by cesarean section to minimize the risk of infection in the newborn.

Note: Women with nongenital lesions can be delivered vaginally, provided the lesions can be covered.

Prevention of herpes simplex virus (HSV) by cesarean section is problematic at this time in most countries with limited resources. This is due in part to:

- lack of recognition of the lesions during or just before labor,
- high rates of home deliveries in most countries,
- limited availability of cesarean sections,
- risk of serious postoperative maternal infection (endometritis and wound infections) following cesarean section, and
- cost issues.

Infants born to mothers with active genital lesions can be cared for in the nursery or NICU, but Standard Precautions, including Transmission-Based (Contact) Precautions, should be used to minimize the risk of transmission to other newborns and health workers. (See **Chapters 2** and **21** for details.) In addition, all waste items (gauze or cotton wet with drainage from the lesions) should be disposed of in a plastic bag or leakproof, covered waste container.

A mother with active lesions (genital or elsewhere) should be placed on Transmission-Based (Contact) Precautions, which includes use of Standard Precautions, until discharged. She should be taught about her infection and measures to prevent postpartum transmission to her baby. Preventive practices the mother should use include:

- Before touching the newborn, the mother should cover nongenital lesions, thoroughly wash and dry her hands and cover her upper body with a clean cloth or gown so that the baby does not come in contact with lesions or potentially infected clothing.
- If the woman has lesions on her lips (cold sores) fever blisters (mouth) or face, she should not cuddle or kiss her baby until the lesions are healed. For added protection and if available, a new disposable paper mask or freshly laundered cloth mask should be worn to cover lips or mouth lesions each time she cares for her infant.
- Breastfeeding can be done provided there are no lesions in the breast area and all skin lesions are covered.
- Direct contact of a newborn with other family members or friends who have active HSV infection should be avoided.

Note: If the mother has genital herpes, her baby may room with her in the hospital if she has been taught the preventive measures.

Human Immunodeficiency Virus

For HIV, the only effective primary prevention is education and counseling as described for HCV above. In areas where HIV prevalence rates are high (>2/1000), pregnant women should be strongly urged to volunteer for counseling and testing. The identification of an HIV-infected pregnant woman as early in the pregnancy as possible is important to ensure appropriate counseling and medical care, including termination of pregnancy if available, and if it is the woman's choice.

HIV can be transmitted from mother to infant in three ways: *in utero* (across the placenta), during delivery or through breastmilk. In breastfed infants, about half the transmission occurs around the time of delivery, one third through breastfeeding and a smaller portion *in utero*. In nonbreastfed infected infants, about two thirds of transmission occurs during or close to delivery and one third *in utero* (DeCock et al 2000). In addition, the risk of HIV transmission from an infected mother to the fetus and infant is affected by other factors as shown in **Table K-2**.

Transmission rates of HIV can be reduced from 25–30% to less than 2% (>90% reduction) if antiretroviral (ARV) drugs are taken in the last

trimester of pregnancy, during labor and delivery, and infants are treated postpartum for 6 weeks as well as not breastfed (Cooper et al 2002). This highly effective regime is not available in developing countries. In a number of countries, however, a shorter and simpler regimen, involving one oral dose of a single ARV drug to the mother during labor and one dose to the newborn within 72 hours, is becoming more widely available. Using this approach, mother-to-infant transmission of the virus can be reduced by nearly 50% (Guay et al 1999).

Table K-2. Factors Associated with Increased Risk of Mother-to-Infant Transmission of HIV

	STRONG EVIDENCE	INTERMEDIATE EVIDENCE	LIMITED EVIDENCE
Maternal Factors	High viral load Immunodeficiency (CD4 <250) HIV infection acquired during pregnancy or breastfeeding	Chorioamnionitis Anemia Sexually transmitted infections Smoking	Frequent unprotected sexual intercourse Vitamin A deficiency Multiple sexual partners Drug use involving injection
Obstetric Factors	Vaginal delivery (compared with elective cesarean section)	Invasive procedures	Episiotomy
Infant Factors	Prematurity Breastfeeding	Lesions of skin and/or mucous membranes (oral thrush)	

Adapted from: McIntyre 2002.

Several studies also have shown that cesarean section before the onset of labor reduces the risk of mother-to-infant transmission. The potential benefit of elective cesarean section, however, has to be balanced against the reported:

- increased risk of postoperative infections in the mother,
- reduced effectiveness in preventing HIV transmission if mothers subsequently breastfeed their infants, and
- limited resources in many developing countries to perform elective cesarean section.

After delivery, care of HIV-infected mothers and infants at risk of infection is the same as for HBV.

Human Papillomavirus

Genital warts caused by HPV, a sexually transmitted virus, are becoming more common. In a small percentage of women, HPV infection is associated with genital cancer (cervix, vagina and vulva), anal cancer in both sexes and penile cancer in men. Primary prevention should involve education and counseling similar to that for HCV, but should reflect the fact that HPV is not transmitted via the blood, but only in vaginal or cervical discharge or through contact with perineal or penile (male) lesions.

There is a small risk that infants born to mothers infected with certain types of HPV may be at increased risk of developing lesions in their respiratory tract (papillomatosis). Because the risk is low, delivery of infected women by cesarean section is not indicated to protect the infant. Cesarean section may be necessary, however, in women whose genital warts are so extensive that soft tissue stretching of the vulva and perineum may not be sufficient to allow vaginal delivery.

Infants born to mothers infected with genital HPV do not need special precautions and can stay in the nursery or NICU.

Rubella

Newborns lacking passively acquired maternal antibodies may develop congenital rubella infection if exposed to the virus during pregnancy. Vaccination of all children and nonpregnant women is the most effective method of preventing congential rubella in infants.[2]

Newborns with congenital rubella infection, or those born to mothers known to have had rubella during pregnancy, should **not** go to the nursery or NICU. They should be placed in a private area. Transmission-Based (Contact) Precautions, including Standard Precautions, should be used to protect other infants and health workers. Where possible, care should be provided to the newborn only by health workers known to have had rubella or those previously vaccinated. Newborns with congenital rubella should be considered contagious for up to 1 year of age.

Pregnant women with active rubella at the time of admission to the hospital should labor and give birth in a separate area. They should be placed in isolation postpartum and be discharged as soon as mother and baby are stabilized. Transmission-Based (Contact) Precautions, including Standard Precautions, should be used to protect other patients in labor or those who are postpartum, as well as susceptible health workers. Where possible, care should be provided to the mother only by health workers known to have had rubella or those previously vaccinated.

Varicella (Chicken Pox)

Newborns lacking passively acquired maternal antibodies may develop a life-threatening infection if exposed to the virus within the last 2 weeks of pregnancy (viral transfer occurs across the placenta) or at the time of

[2] Vaccinated women should be counseled to avoid pregnancy for 3 months because of the possible small risk the vaccine could cause a congenital abnormality.

delivery. The greatest risk is if the baby is born within 2 days before or 5 days after the onset of maternal chicken pox. Infants at risk should receive varicella immune globulin, 1.25 mL (one vial) intramuscularly. In addition, the newborn should be placed in isolation to minimize the risk of transmission (airborne) to other newborns and susceptible postpartum mothers and healthcare staff. Where possible, care should be provided to the newborn only by health workers known to have had varicella or those previously vaccinated.

Pregnant women with active varicella at the time of admission to the hospital should labor and give birth in a separate area. They should be placed in isolation postpartum and discharged as soon as mother and baby are stabilized. In addition, their newborns should be separated from them until all lesions are healed. Transmission-Based (Droplet and Contact) Precautions, including Standard Precautions, should be used to protect other patients in labor or who are postpartum as well as susceptible health workers. Where possible, care should be provided to the mother only by health workers known to have had varicella or those previously vaccinated.

REFERENCES

American Academy of Pediatrics (AAP) and American Academy of Obstetricians and Gynecologists (ACOG). 1997. *Guidelines for Perinatal Care*, 4th ed, revised. AAP: Elk Grove, IL.

Centers for Disease Control and Prevention (CDC). 2000. Hospital-based policies for prevention of perinatal group B streptococcal disease. *MMWR* 49(41): 936–940.

Centers for Disease Control and Prevention (CDC). 1996. Prevention of perinatal group B streptococcal infections disease: A public health perspective. *MMWR* 45(RR-7): 1–24.

Cooper ER et al. 2002. Combination antiretroviral strategies for the treatment of pregnant HIV-1 infected women and prevention of perinatal HIV-1 transmission. *J Acquir Immune Defic Syndr* 29(5): 484–494.

DeCock KM et al. 2000. Prevention of mother-to-child HIV transmission in resource-poor countries: Translating research into policy and practice. *JAMA* 283(9): 1175–1182.

Guay LA et al. 1999. Intrapartum and neonatal single-dose nevirapine compared with zidovudine for prevention of mother-to-child transmission of HIV-1 in Kampala, Uganda: HIVNET 012 randomized trial. *Lancet* 354(9181): 795–802.

Keenan C. 1998 Prevention of neonatal group B streptococcal infection. *Am Fam Physician* 57(11): 2713–2725.

McIntyre JA. 2002. Mother-to-child transmission of HIV-1. *IPPF Medical Bulletin* 36(4): 1–2.

GLOSSARY

Airborne transmission: Transfer of particles 5 μm or less in size into the air, either as airborne droplets or dust particles containing the infectious microorganism; can be produced by coughing, sneezing, talking or procedures such as bronchoscopy or suctioning; can remain in the air for up to several hours; and can be spread widely within a room or over longer distances. Special air handling and ventilation are needed to prevent airborne transmission. (**Chapter 21 and Appendix I**)

Amphoteric: Organic chemical (e.g., amino acid) having both acid and basic properties.

Animate: Property of having life or being alive (e.g., human tissue or organs).

Anionic: Positively charged particle or substance (i.e., in electrolysis, anions move toward the negatively charged cathode); opposite of cationic.

Antisepsis: Process of reducing the number of microorganisms on skin, mucous membranes or other body tissue by applying an antimicrobial (antiseptic) agent. (**Chapters 1, 6 and 23**)

Antiseptic or **antimicrobial agent (terms used interchangeably)**: Chemicals that are applied to the skin or other living tissue to inhibit or kill microorganisms (both transient and resident) thereby reducing the total bacterial counts. (**Chapters 6 and 23**)

Antiseptic handrub or **waterless, alcohol-based antiseptic handrub (terms used interchangeably)**: Fast acting antiseptic handrubs that do not require use of water to remove transient flora, reduce resident microorganisms and protect the skin. Most contain 60–90% alcohol, an emollient and often an additional antiseptic (e.g., 2–4% chlorhexidine gluconate) that has residual action. (**Chapter 3 and Appendix B**)

Asepsis and **aseptic technique**: Combination of efforts made to prevent entry of microorganisms into any area of the body where they are likely to cause infection. The goal of asepsis is to **reduce to a safe level** or **eliminate** the number of microorganisms on both animate (living) surfaces (skin and tissue) and inanimate objects (surgical instruments and other items). (**Chapters 1 and 7**)

At point of use: Equipment, instruments and supply items are at the place where needed (e.g., sharps containers are placed within an arm's reach of where injections are being given). (**Chapter 15**)

Autoclave: Device that sterilizes instruments or other objects by using steam under pressure. The length of time required for sterilization depends on temperature and pressure. (**Chapter 11 and Appendix G**)

Bacterial endotoxins: Lipopolysaccharide components from gram-negative bacteria cell membranes that result from bacterial metabolism. Endotoxins survive sterilization because they require dry heat at 270° F (132°C) for 1 hour to be inactivated. They can cause pyrogenic reaction symptoms, including fever, chills, and hypertension.

Bactericide: Agent that kills bacteria.

Bioburden: Number of types of viable microorganisms with which an item is contaminated; also known as ***bioload*** or ***microbial load***.

Biological indicator: Sterilization process monitoring device consisting of a standardized, viable population of microorganisms (usually bacterial spores) known to be resistant to the process of sterilization being monitored. Biological indicators are intended to demonstrate whether or not the conditions were adequate to achieve sterilization. A negative biological indicator does not prove that all items in the load are sterile or that they were all exposed to adequate sterilization conditions. (**Chapter 11 and Appendix G**)

Biosafety level (BSL) guidelines: Combination of primary and secondary containment and safety guidelines designed for use in microbiology laboratories and bacteriology research units functioning at four levels (BSL-1 to BSL-4) of increasing risk. (**Chapter 17**)

Biological safety cabinets (BSCs): Devices that provide protection for personnel, the agent being processed and the environment. They range in complexity from level I (general research cabinets for use with low- to moderate-risk microorganisms) to level III (totally enclosed cabinets with gas-tight construction that provide maximum protection to workers and the environment). (**Chapter 17**)

Blood bank: Facility or hospital unit that performs the collection, processing, storage and distribution of human blood or blood products. (**Chapter 18**)

Bowie-Dick test: Diagnostic test of a sterilizer's ability to remove air from the chamber of a prevacuum steam sterilizer. The air-removal or Bowie-Dick test is not a test for sterilization. (**Appendix G**)

Casefinding: Method of identifying patients with nosocomial infections through a combination of: 1) reviewing medical records, 2) asking questions directed to patients or health workers and, 3) checking laboratory, X-ray or other relevant data if available. (**Chapter 28**)

Cationic: Negatively charged particle or substance (i.e., in electrolysis, cations move toward the positively charged anode); opposite of anionic.

Chemical indicator: Sterilization process monitoring device designed to respond with a characteristic chemical change to one or more of the physical conditions within the sterilizing chamber. Chemical indicators are intended to detect potential sterilization failures that could result from incorrect packaging, incorrect loading of the sterilizer, or malfunctions of the sterilizer. The "pass" response of a chemical indicator does not prove that the item accompanied by the indicator is sterile. (**Chapter 11 and Appendix G**)

Clean water: Natural or chemically treated and filtered water that is safe to drink and use for other purposes (e.g., handwashing and medical instrument cleaning) because it meets specified public health standards. These standards include: zero levels of microorganisms, such as bacteria (e.g., fecal coliform and *Escherichia coli*), parasites (e.g., *Giardia lamblia*) and viruses (e.g., hepatitis A or E); low turbidity (cloudiness due to particulate matter and other contaminants); and minimum levels of disinfectants, disinfectant by-products, inorganic and organic chemicals and radioactive materials. At a minimum clean water should be free of microorganisms and have low turbidity (is clear, not cloudy). (**Chapters 3 and 26**)

Cleaning: Process that physically removes all visible dust, soil, blood or other body fluids from inanimate objects as well as removing sufficient numbers of microorganisms to reduce risks for those who touch the skin or handle the object. (**Chapters 1, 9 and 10**)

Cleaning solution: Any combination of soap (or detergent) and water used to wash or wipe down environmental surfaces such as floors, walls, ceilings and furniture. (**Chapter 6**)

Clinically significant antibody: Antibody capable of producing an adverse reaction to transfused blood or blood product obtained from a donor (allogenic antibody) or recipient (autologous antibody). (**Chapter 18**)

Closed system for obtaining blood: System in which the blood is not exposed to air or outside elements during collection, processing—including separation of components (e.g., platelets) if required prior to transfusion—and storage. It is the safest way to collect, process and store blood. (**Chapter 18**)

Cohorting: Practice of placing patients with the same active infectious disease (e.g. chicken pox)—but no other infection—in the same room or ward. (**Chapter 21**)

Colonization: Pathogenic (illness- or disease-causing) organisms are present in a person (i.e., they can be detected by cultures or other tests) but are not causing symptoms or clinical findings (i.e., no cellular changes or damage). (**Chapters 1 and 21**)

Contact time: Amount of time a disinfectant is in direct contact with the surface or item to be disinfected. For surface disinfection, this time period starts with the application to the surface, and ends when complete drying has occurred. (**Appendix F**)

Contact transmission: Infectious agent (bacteria, virus or parasite) transmitted directly or indirectly from one infected or colonized person to a susceptible host (patient), often on the contaminated hands of a health worker. (**Chapter 21**)

Container: Vessel in which waste is placed for handling, transportation, storage and/or eventual disposal. (**Chapter 8**)

Contaminated: State of having been actually or potentially in contact with microorganisms. As used in healthcare, the term generally refers to the presence of microorganisms that could be capable of producing disease or infection. (**Chapters 8 and 20**)

Corrosion: Action of chemical solutions, such as those containing salt (sodium chloride) or commercial bleach (sodium hypochlorite at concentrations above 0.5%), to cause metal instruments to be gradually eaten away (rusted) with prolonged contact (i.e., more than 1 hour). (**Appendix E**)

Critical medical device (or item): Devices that penetrate skin or invade normally sterile parts of the body (e.g., central venous catheters). These items contact blood and require sterilization. (**Chapter 1**)

Culture: Growth of microorganisms in or on a nutrient medium; to grow microorganisms in or on such a medium.

Culture medium: Laboratory substance or preparation used to grow and cultivate microorganisms.

Decontamination: Process that makes inanimate objects **safer** to be handled by staff **before** cleaning (i.e., inactivates HBV, HCV and HIV and reduces, but does not eliminate, the number of other contaminating microorganisms). (**Chapters 1, 9 and 10 and Appendices E, F and H**)

Detergents and **soaps (terms used interchangeably)**: Cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt and debris and transient microorganisms from hands. **Plain** soaps require friction (scrubbing) to mechanically remove microorganisms while **antiseptic** (antimicrobial) soaps also kill or inhibit growth of most microorganisms. (**Chapters 3, 13 and 16 and Appendix B**)

Disinfectant: Chemical that destroys or inactivates microorganisms. Disinfectants are classified as low-, intermediate- or high-level depending on their ability to kill or immobilize some (low- or intermediate-level) or all (high-level) microorganisms (but not all spores). Phenols, chlorine or chlorine-containing compounds and quaternary ammonium compounds (QUATs) are classes of disinfectants frequently used to clean noncritical surfaces such as floors, walls and furniture. (**Chapters 12 and 16 and Appendix F**)

Disinfectant cleaning solution: Products that are a combination of a detergent (soap) and a chemical disinfectant. Not all detergents and disinfectants are compatible. Several combinations are available commercially or can be prepared, such as alkaline detergents with chlorine compounds, alkaline detergents with QUATs or other nonionic surfactants, and acid detergents with iodophors. (**Chapter 16 and Appendix F**)

Disposal: Intentional burial, deposit, discharge, dumping, placing or release of any waste material into or on air, land or water. Disposal is undertaken without the intention of retrieval. (**Chapter 8**)

Donor-Patient: Person whose blood is collected for possible transfusion to another person (allogenic transfusion). (**Chapter 18**)

Donor-Recipient: Person whose own blood is collected for possible transfusion to her/himself (autologous transfusion). (**Chapter 18**)

Droplet transmission: Contact of the mucous membranes of the nose, mouth or conjunctivae of the eye with infectious particles larger than 5 μm in size that can be produced by coughing, sneezing, talking or procedures such as bronchoscopy or suctioning. Droplet transmission requires close contact between the source and the susceptible person because particles remain airborne briefly and travel only about 1 meter (3 feet) or less. (**Chapter 21**)

Dry heat sterilization: Oven that sterilizes metal instruments, glass syringes and bottles and other items by dry heat. Plastic and rubber items cannot be dry-heat sterilized because temperatures used (160–170° C) are too high for these materials. (**Chapter 11 and Appendix G**)

Electrolyte: Chemical substance, such as salt (sodium chloride) or commercial bleach (sodium hypochlorite), which in solution separates into positive (sodium) and negative (chloride or hypochlorite) ions and is capable of conducting an electric current by movement of the positive and negative ions to oppositely charged electrodes. (**Appendix F**)

Electrolytic corrosion: Chemical process that occurs when two or more different types of metal (e.g., stainless steel medical instruments in an aluminum pan) are placed in water or weak salt (sodium chloride) or commercial bleach (sodium hypochlorite) solution. (To avoid this, instruments should be placed in plastic pans and instruments made of different metals should not be allowed to touch each other.) (**Appendix E**)

Emollient: Organic liquid, such as glycerol, propylene glycol or sortibol, that when added to handrubs and hand lotions softens the skin and helps prevent skin damage (cracking, drying, irritation and dermatitis) due to frequent handwashing with soap (with or without antiseptic agent) and water. (**Chapter 3**)

Encapsulation: Filling a sharps container that is three-quarters full with cement or clay, which, after hardening, can be disposed of safely in a landfill. (**Chapter 8**)

Endemic illness or disease: Infectious disease, such as cholera or AIDS, which is continuously present at some level (prevalence) in a particular country or region. (**Chapter 26**)

Endometritis: Acute postpartum infection of the lining (endometrium) of the uterus with extension into the smooth muscle wall (myometrium). Clinical features include fever, usually developing on the first or second postpartum day, uterine tenderness, lower abdominal pain, foul-smelling vaginal discharge (lochia) and signs of peritonitis in women who have had a cesarean section. (**Chapter 25**)

Endospore or **spore (terms used interchangeably)**: Relatively water-poor round or elliptical resting cell consisting of condensed cytoplasm and nucleus surrounded by an impervious cell wall or coat. Spores are relatively resistant to disinfectants and sterilants, specifically the bacillus and clostridium species. (**Chapters 1, 9 and 12 and Appendix F**)

Environmental controls: Standards specifying procedures to be followed for the routine care, cleaning and disinfection of environmental surfaces, beds, bedrails, bedside equipment and other frequently touched surfaces. (**Chapters 15 and 16**)

Environmental hygiene: Process of maintaining a clean, healthy and pleasing patient and work environment. (**Chapter 16**)

Epidemic: Rapid spread of an infectious disease, such as cholera, among many individuals in a hospital or community at the same time. (**Chapter 26**)

Episiotomy: Surgical cut made in the perineum (usually at the 6 o'clock position) just prior to delivery. The purpose is to facilitate delivery of the presenting part and minimize the risk of injury to the perineal area. Episiotomies are, however, associated with increased bleeding, may lead to increased tearing (3rd or 4th degree perineal laceration), frequently become infected and, most importantly, usually not necessary. (**Chapter 25**)

Exit site infection (microbiologic diagnosis): Clinical infection in which culture of the discharge (pus or fluid) at the exit site yields a microorganism, with or without microbiologic evidence of bloodstream infection. (**Chapter 23**)

Exposure time: Period of time during a sterilization process in which items are exposed to the sterilant at the specified sterilization parameters. In a steam sterilization process, exposure time is the period during which items are exposed to saturated steam at the specified temperature. (**Appendix G**)

Flash sterilization: Process designed for the steam sterilization of patient-care items for immediate use. (**Appendix G**)

Handwashing: Process of mechanically removing soil and debris from the skin of hands using plain soap and water. (**Chapter 3 and Appendices A and B**)

Hazard: Intrinsic potential property or ability of any agent, equipment, material or process that can cause harm. (**Chapter 8**)

High-level disinfection (HLD): Process that eliminates **all** microorganisms **except some** bacterial endospores from inanimate objects by boiling, steaming or the use of chemical disinfectants. (**Chapters 1, 9 and 12 and Appendix F**)

Hospital-acquired infection or **nosocomial (terms used interchangeably)**: Infection that is neither present nor incubating at the time the patient came to the hospital. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.) (**Chapters 3, 20 and 21**)

Hydrophilic (water-seeking): Substances that are capable of dissolving in, taking up or being attracted to water but **not** fats or oils (synonym: lipophobic).

Hydrophobic (water-avoiding): Substances that are **not** capable of dissolving in, taking up or being attracted to water but are attracted to fats and oils (synonym: lipophilic).

Inanimate: Object or article (e.g., surgical instrument, gloves or other items) that does not have life (not animate).

Inanimate surface: Nonliving surface (e.g., floors, walls, furniture).

Incineration: Controlled burning of solid, liquid or gaseous combustible (burnable) wastes to produce gases and residues containing little or no burnable material. (**Chapter 8**)

Infectious microorganisms: Microorganisms capable of producing disease in appropriate hosts.

Infectious waste: The part of medical waste that is capable of causing infectious diseases. (**Chapter 8**)

Intermediate-level disinfectant: Agent that destroys all vegetative bacteria, including tubercle bacilli, lipid and some nonlipid viruses, and fungus spores, but not bacterial spores. (**Appendix F**)

Intra-amniotic infection syndrome (IAIS), also referred to as amnionitis or chorioamnionitis: Acute clinically detectable infection in the uterus and its contents (fetus, placenta and amniotic fluid) during pregnancy. (**Chapter 25**)

Invasive group B streptococcal sepsis: Newborn infection characterized by bacteremia, pneumonia, meningitis and death in up to 25% of infants with the infection. It occurs most commonly following IAIS. Other sites of infection include newborn skin infections (cellulitis) and infections in bones (osteomyelitis). (**Chapter 25**)

Laboratory-acquired infection: Nosocomial infection resulting from the performance of laboratory activities by staff, regardless of how it occurred. (**Chapters 17 and 20**)

Linens: Cloth items used in healthcare facilities by housekeeping staff (bedding and towels), cleaning staff (cleaning cloths, gowns and caps) and surgical personnel (caps, masks, scrub suits, surgical gowns, drapes and wrappers) as well as by staff working in specialty units such as ICUs and other units performing invasive medical procedures (e.g., anesthesiology, radiology or cardiology). (**Chapters 5 and 13**)

Lipid virus: Microorganism consisting of a nucleic acid core surrounded by a protein coat and a lipoprotein envelope. This type of virus (e.g., HIV) generally is easily and rapidly inactivated by most disinfectants (e.g., dilute chlorine solutions). Also referred to as an enveloped or lipophilic (fat-seeking) virus.

Lipophilic (fat-seeking): Substances that are capable of dissolving in, taking up or being attracted to fats or oils but are **not** attracted to water (synonym: hydrophobic).

Lipophobic (fat-avoiding): Substances that are not capable of dissolving in, taking up or being attracted to fats but are attracted to water (synonym: hydrophilic).

Lookback systems: Process of identifying persons who have received blood transfusions from donors who are subsequently found to have infections with HCV, HIV (and often HBV), and notifying them if appropriate. (**Chapter 18**)

Low-level disinfectant: Agent that destroys all vegetative bacteria (except tubercle bacilli), lipid viruses, some nonlipid viruses, and some fungus, but not bacterial spores. (**Appendix F**)

Mechanical indicator: Automated devices that monitor the sterilization process (e.g., graphs, gauges, printouts). (**Chapter 11 and Appendix G**)

Microorganism: Causative agent of infection. They include bacteria, viruses, fungi and parasites. For infection prevention purposes, bacteria can be further divided into three categories: vegetative (e.g., staphylococcus), mycobacteria (e.g., tuberculosis) and endospores (e.g., tetanus). Of all the common infectious agents, endospores are the most difficult to kill due to their protective coating. (**Chapter 1**)

Minimum effective concentration (MEC): Minimum concentration of a chemical disinfectant needed to achieve the claimed microbicidal (killing) activity as determined by dose-response testing.

Municipal waste: General waste for collection by municipalities (e.g., local city or town authorities) generated mainly by households, commercial activities and street sweeping. (**Chapter 8**)

Muslin: Moderately woven, 100% cotton cloth. (**Chapter 5**)

Mycobacteria: Bacteria with a thick, waxy coat that makes them more resistant to chemical disinfectants than other types of vegetative bacteria.

Noncritical medical device (or item): Devices that normally make contact with the patient's intact skin (e.g., blood pressure cuff, oxygen masks). These devices require low- to intermediate-level disinfection, and reuse carries little risk. (**Chapter 1**)

Nonionic: Neutral (neither positively or negatively charged) particle or substance.

Nonlipid virus: Virus that is more resilient to inactivation than a lipid virus. Nonlipid viruses are also referred to as nonenveloped or hydrophilic (water-seeking) viruses.

Nosocomial or **hospital-acquired infection (terms used interchangeably)**: Infection that is neither present nor incubating at the time the patient came to the hospital. (Nosocomial refers to the association between care and the subsequent onset of infection. It is a time-related criterion that does not imply a cause and effect relationship.) (**Chapters 3, 20, 21 and 28**)

Nosocomial diarrhea: On at least 2 consecutive days having at least three loose or watery stools with the onset more than 72 hours after admission to the hospital (or more days than the incubation period if the agent is known). (**Chapter 26**)

Nosocomial infection in newborns: Infection occurring after birth but excluding those infections known to have been transmitted across the placenta such as congenital syphilis, cytomegalovirus, rubella, varicella (chicken pox) and the protozoan parasite, *Toxoplasmosis gondii*. (**Chapter 25**)

Nosocomial infection in obstetrical patients: Infection that is neither present nor incubating at the time the patient is admitted to the hospital. Most urinary tract infections and endometritis are nosocomial even though the causative organism may be endogenous (i.e., present in the maternal lower genital tract prior to delivery). (**Chapter 25**)

Occupational injury or **infection**: Injury or infection acquired by healthcare staff while performing their normal duties. (**Chapters 13 and 20**)

Operating room: Area or space where surgical procedures are performed. (**Chapter 15**)

Organ/Space SSI: Any part of the body other than the incised body wall parts that were opened or handled during an operation. (**Chapter 23**)

Parts per million (ppm): Concentrations of trace contaminant gases in the air (or chemicals in a liquid) are commonly measured in parts per million (ppm) by volume. To convert percent concentration to ppm and vice versa, use this formula: ppm = percent (%) x 10,000. (**Chapter 26**)

Pasteurization: Process developed by Louis Pasteur of heating milk, wine, or other liquids to 60°C to 100°C (or the equivalent) for approximately 30 minutes to kill or markedly reduce the number of pathogenic and spoilage organisms other than bacterial spores. (**Chapter 12**)

Personal protective equipment (PPE): Specialized clothing or equipment (e.g., gloves, face mask or plastic apron) worn by an employee for protection against exposure to blood or body fluids or other hazards. Uniforms, pants, and shirts not designed to function as protection against a hazard are not considered to be PPE. (**Chapter 5**)

Phlebitis: Area of swelling, redness, warmth and tenderness of the skin around the site where the intravascular catheter comes out of the skin (the exit site). If phlebitis is associated with other signs of infection, such as fever and pus coming from the exit site, it is classified as a **clinical exit site infection**. (**Chapter 23**)

Pocket infection: Infected fluid isolated from the area around a totally implanted intravascular device, with or without microbiologic evidence of bloodstream infection. (**Chapter 24**)

Prions: Protein-containing infectious agents that require special handling and processing because they are resistant to heat and high-pressure steam sterilization. Prions are the only known infectious agents that do not contain either DNA or RNA. (**Chapter 11**)

Protective barrier: Physical, mechanical or chemical process that helps prevent the spread of infectious microorganisms from person to person (patient, healthcare client or health worker), and from equipment, instruments and environmental surfaces to people. (**Chapter 1**)

QUAT: Abbreviated form of the term quaternary ammonium compound; a surface-active, water-soluble low-level disinfecting substance that has four carbon atoms linked to a nitrogen atom through chemical (covalent) bonds. (**Chapter 16 and Appendices B and F**)

Recipient transfusion reaction: Adverse reaction to infusing blood or blood products into a patient (recipient). (**Chapter 18**)

Recycling: Physical and/or chemical process that recovers the basic material in a product (e.g., paper from newspapers, aluminum from soft drink cans or plastic from disposable syringes) for reuse as a new or different product. (**Chapter 14**)

Reprocessing: Decontaminating, disassembling (if necessary), cleaning, inspecting, testing, packaging, relabeling, and sterilizing or high-level disinfecting single-use devices (SUDs) after they have been used on a patient for their intended purpose. Reprocessing also is performed on SUDs that were removed from the package (or container) but not used on a patient or whose expiration date has passed. (**Chapter 14**)

Resident flora: Microorganisms that live in the deeper layers of the skin, as well as within hair follicles, and cannot be completely removed, even by vigorous washing and rinsing with plain soap and clean water. (**Chapter 3**)

Resterilization: Repeat application of a terminal process designed to remove or destroy all viable forms of microbial life, including bacterial spores, to an acceptable sterility assurance level. This process is performed on devices whose expiration date has passed or that have been opened and may or may not have been used on a patient. (**Chapters 11 and 14**)

Safe Zone (also Neutral Zone): Device or designated area of the sterile field in which sharps are placed, accessed, returned, and retrieved to avoid hand-to-hand transfer of sharps between personnel. (**Chapter 7**)

Sanitary landfill: Engineered method of disposing of solid waste on land in a manner that protects the environment (e.g., by spreading the waste in thin layers, compacting it to the smallest practical volume and then covering it with soil at the end of each working day). (**Chapter 8**)

Sanitizer: Chemical that reduces the number of bacterial contaminants to safe levels on inanimate objects based on public health requirements (i.e., a chemical that kills 99.999% of the specific test bacteria in 30 seconds under the conditions of the test). (**Chapter 16**)

Scavenging: Manual sorting of solid waste at landfills and removal of usable material. (**Chapter 8**)

Segregation: Systematic separation of solid waste into designated categories. (**Chapter 8**)

Semicritical medical device (or item): Devices that come in contact with mucous membranes or nonintact skin during use (e.g., endoscopes, respiratory equipment). These devices require high-level disinfection if sterilization is not practical, and reuse carries a greater risk for cross-contamination than noncritical items. (**Chapters 1, 9 and 12 and Appendices F and H**)

Septic pelvic thrombophlebitis: Thrombosis (blockage) of the deep pelvic veins due to inflammation and blood clots. It is uncommon (approximately 1 in 2000 deliveries). Predisposing factors include cesarean section after long labor (>24 hours), premature rupture of membranes, difficult delivery (forceps or vaginal extraction), anemia and malnutrition. (**Chapter 25**)

Sewerage: System for the collection and transport of sewage, including conduits, pipes and pumping stations. (**Chapter 8**)

Sharps: Suture needles, scalpel blades, scissors, wire sutures, broken glass or any object that can cause a puncture or cut. (**Chapters 7 and 8 and Appendix E**)

Soaps and **detergents (terms used interchangeably)**: Cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands. **Plain** soaps require friction (scrubbing) to mechanically remove microorganisms while **antiseptic** (antimicrobial) soaps also kill or inhibit growth of most microorganisms. (**Chapters 3, 13 and 16 and Appendix B**)

Soiled or **contaminated linen**: Linen from multiple sources within the hospital or clinic that has been collected and brought to the laundry for processing. All items, regardless of whether or not they are visibly dirty or have been used in a surgical procedure, must be washed and dried. (**Chapter 13**)

Sorting: Process of inspecting and removing foreign, and in some cases dangerous, objects (e.g., sharps or broken glass), from soiled linen before washing. This step is extremely important because soiled linen from the operating room or clinic occasionally contains sharps (e.g., scalpels, sharp-tipped scissors, hypodermic and suture needles and towel clips). (**Chapter 13**)

Spaulding classification: Strategy for reprocessing contaminated medical devices. The system classifies medical devices as critical, semicritical, or noncritical based upon the risk from contamination on a device to patient safety. (**Chapters 1 and 15**)

Spore or **endospores (terms used interchangeably)**: Relatively water-poor round or elliptical resting cell consisting of condensed cytoplasm and nucleus surrounded by an impervious cell wall or coat. Spores are relatively resistant to disinfectants and sterilants, specifically the bacillus and clostridium species. (**Chapters 1, 9 and 12 and Appendix F**)

Steam sterilization: Sterilization process that uses saturated steam under pressure, for a specified exposure time and at a specific temperature, as the sterilizing agent. (**Chapter 11 and Appendix G**)

Sterilants: Chemicals used to destroy all forms of microorganisms, including endospores. Most sterilants are also high-level disinfectants when used for a shorter period of time. Sterilants are only used on inanimate objects (e.g., surgical instruments) that are used in semicritical and critical areas (e.g., surgery). Sterilants are not meant to be used for cleaning environmental surfaces. (**Chapters 9 and 11 and Appendix F**)

Sterile or **sterility**: State of being free from all living microorganisms. In practice, usually described as a probability function (e.g., the probability of a microorganism surviving sterilization as being one in a million). (**Chapter 12 and Appendices F and G**)

Sterilization: Process that eliminates **all** microorganisms (bacteria, viruses, fungi and parasites) **including** bacterial endospores from inanimate objects by high-pressure steam (autoclave), dry heat (oven), chemical sterilants or radiation. (**Chapters 1, 9 and 11 and Appendix G**)

Sterilizer: Apparatus used to sterilize medical instruments, surgical gloves, equipment or supplies by direct exposure to the sterilizing agent (autoclave or dry-heat oven). (**Chapter 11 and Appendix G**)

Surfactant: Agent that reduces the surface tension of water or the tension at the interface between water and another liquid; a wetting agent found in many sterilants and disinfectants. (**Chapter 16**)

Surgical asepsis: Preparation and maintenance of a reduced (safe) level of microorganisms during an operation by controlling four main sources of infectious organisms: the patient, personnel, equipment and the environment. (**Chapters 6, 7 and 23**)

Surgical site infections (SSI): Either an **incisional** or **organ/space** infection occurring within 30 days after an operation or within 1 year if an implant is present. Incisional SSIs are further divided into **superficial incisional** (only involves skin and subcutaneous tissue) and **deep incisional** (involves deeper soft tissue, including fascia and muscle layers). (**Chapter 23**)

Surgical unit: Whole surgical area including lockers and dressing rooms, preoperative and recovery rooms, peripheral support areas including storage space for sterile and high-level disinfected items and other consumable supplies, corridors leading to restricted areas, the operating room(s), scrub sink areas and the nursing station. (**Chapters 7 and 15**)

Surveillance: Systematic collection of relevant data on patient care, the orderly analysis of the data and the prompt reporting of the data to those who need it. **Active surveillance** consists of collecting information directly from patients or staff, while **passive surveillance** includes examining reports, laboratory information and data from other sources. (**Chapter 28**)

Time-weighted average (TWA): Average of all concentrations of a chemical to which a worker has been exposed during a specific sampling time, reported as an average over the sampling time. The permissible exposure limit for ethylene oxide is 1 ppm as an 8-hour TWA. Exposures above the ppm limit are permitted if they are compensated for by equal or longer exposures below the limit during the 8-hour workday. (**Appendix F**)

Transfusion service: Facility or hospital unit that provides storage, pretransfusion testing and cross-matching, and infusion of blood or blood products to intended patients (recipients). (**Chapter 18**)

Transient flora: Microorganisms acquired through contact with patients, other healthcare workers or contaminated surfaces (e.g., examination tables, floors or toilets) during the course of the normal workday. These organisms live in the upper layers of the skin and are partially removed by washing with plain soap and clean water. (**Chapter 3**)

Tunnel infection: Tenderness, redness and swelling for more than 2 cm (about 1 inch) along the tract of an intravascular catheter, with or without microbiologic evidence of local or bloodstream infection. (**Chapter 24**)

Type of detergent: Commercial cleaning product (liquid or powder) that are composed of a hydrophilic (water-seeking) component and a lipophilic (fat-seeking) component and can be divided into four types: anionic, cationic, amphoteric and nonionic detergents. (**Chapter 16**)

Unit of blood: Sterile plastic bag in which a fixed volume of blood is collected in a suitable amount of anticoagulant. (**Chapter 18**)

Urticarial reaction: Allergic reaction consisting of itching (pruritis), hives, skin rash and/or similar allergic condition occurring during or following a transfusion of blood or blood products. (**Chapter 18**)

Vegetative bacteria: Bacteria that are devoid of spores and usually can be readily inactivated by many types of germicides.

Visibly soiled hands: Hands showing visible dirt or are visibly contaminated with blood or body fluids (urine, feces, sputum or vomit). (**Chapter 3**)

Waste management: All activities, administrative and operational (including transportation activities), involved in the handling, treatment, conditioning, storage and disposal of waste. (**Chapter 8**)

Waterless, alcohol-based antiseptic handrub or **antiseptic handrub (terms used interchangeably)**: Fast acting antiseptic handrubs that do not require use of water to remove transient flora, reduce resident microorganisms and protect the skin. Most contain 60–90% alcohol, an emollient and often an additional antiseptic (e.g., 2–4% chlorhexidine gluconate) that has residual action. (**Chapter 3 and Appendix B**)